PB82-217795

Development of Techniques and Methodology for the Laboratory Culture of Striped Bass, Morone Saxatilis (Walbaum)

Rhode Island Univ., Kingston

Prepared for

Environmental Research Lab.
Narragansett, RI

Apr 82

U.S. Department of Commerce
National Technical Information Service
NTIS

April 1982
EPA-600/3-82-017

DEVELOPMENT OF TECHNIQUES AND METHODOLOGY FOR THE LABORATORY CULTURE OF STRIPED BASS, *MORONE SAXATILIS* (WALBAUM)

PB82-217795

by

Bruce A. Rogers, Deborah T. Westin
and
Saul B. Saila

Graduate School of Oceanography
University of Rhode Island
Kingston, Rhode Island 02881

Grant No. 68-03-0316

Project Officer

Allan D. Beck
Environmental Research Laboratory
Narragansett, Rhode Island 02882

ENVIRONMENTAL RESEARCH LABORATORY
OFFICE OF RESEARCH AND DEVELOPMENT
U.S. ENVIRONMENTAL PROTECTION AGENCY
NARRAGANSETT, RHODE ISLAND 02882

REPRODUCED BY
NATIONAL TECHNICAL
INFORMATION SERVICE
U.S. DEPARTMENT OF COMMERCE
SPRINGFIELD, VA. 22161

TECHNICAL REPORT DATA

(Please read instructions on the reverse before completing)

1. REPORT NO. EPA-600/3-82-018

2. ORD Report

3. RECIPIENT'S ACCESSION NO. PB82 217795

4. TITLE AND SUBTITLE Development of Techniques and Methodology for the Laboratory Culture of Striped Bass, Morone saxatilis.

5. REPORT DATE March 1982

6. PERFORMING ORGANIZATION CODE

7. AUTHOR(S) Bruce A. Rogers, Deborah T. Westin, and Saul B. Saila

8. PERFORMING ORGANIZATION REPORT NO. ERLN-X2

9. PERFORMING ORGANIZATION NAME AND ADDRESS
Graduate School of Oceanography, University of Rhode Island, Kingston, RI, 02881

10. PROGRAM ELEMENT NO. ACUL1A

11. CONTRACT/~~GRANT~~ NO. Contract 68-03-0316

12. SPONSORING AGENCY NAME AND ADDRESS
U. S. Environmental Protection Agency
ERL, Narragansett
South Ferry Road
Narragansett, RI, 02882

13. TYPE OF REPORT AND PERIOD COVERED Final Contract Report

14. SPONSORING AGENCY CODE EPA/600/05

15. SUPPLEMENTARY NOTES This project has been financed, in part, with funds from the U. S. Environmental Protection Agency.

16. ABSTRACT

This report describes the research undertaken to develop laboratory culture techniques for striped bass (Morone saxatilis) which can be used to provide an adequate supply of various life stages of this important fish species for water quality and hazard evaluation testing. For each of the four life stages (egg, larva, juvenile and adult), the upper and lower lethal levels, where applicable, and an approximation of optimum conditions were defined with regard to physical characteristics of the environment including temperature, salinity, dissolved oxygen, light, and turbidity. Satisfactory laboratory diets were defined and verified for each life stage. A comprehensive set of procedures was developed and descrived in a step-by-step manner for use by research personnel wishing to maintain laboratory populations of striped bass for physiological and toxicological use.

17. KEY WORDS AND DOCUMENT ANALYSIS

a. DESCRIPTORS	b. IDENTIFIERS/OPEN ENDED TERMS	c. COSATI Field/Group

18. DISTRIBUTION STATEMENT Distribution Unlimited

19. SECURITY CLASS *(This Report)* Unclassified

20. SECURITY CLASS *(This page)* Unclassified

21. NO. OF PAGES

22. PRICE

EPA Form 2220-1 (9-73)

DISCLAIMER

This report has been reviewed by the Environmental Research Laboratory, Narragansett, U.S. Environmental Protection Agency, and approved for publication. Approval does not signify that the contents necessarily reflect the views and policies of the U.S. Environmental Protection Agency, nor does mention of trade names or commercial products constitute endorsement or recommendation for use.

FOREWORD

The Narragansett Environmental Research Laboratory of the U. S. Environmental Protection Agency is conducting research to develop a firm, factual basis for regulatory decisions designed to protect man and his environment from damaging effects of pollutants in marine waters. Information needed in such decisions includes:

- Physical and chemical fate and transformations of pollutants in marine systems
- Responses of marine organisms to pollutant stress
- The resultant effects of different organism and community responses on the integrity of ecosystems and biogeochemical processes

Measurements of biological effect have often suffered from a lack of reproducibility and of comparability of results between investigators. This report presents standard methods for obtaining test organisms in more uniform physiological condition, and thus should contribute to increased reliability of test results.

Tudor T. Davies
Director

ABSTRACT

This research was undertaken to develop laboratory culture techniques for striped bass (*Morone saxatilis*) that could be used to provide an adequate supply of various life stages of this important fish species for water quality and hazard evaluation testing. The work included both an extensive literature review of the data available on all aspects of its life history and a program of laboratory experiments to determine the optimal rearing conditions for each life stage.

For each of the four life stages defined here, egg, larva, juvenile, and adult, the upper and lower lethal levels where applicable and an approximation of optimum conditions were defined with regard to physical characteristics of the environment including temperature, salinity, dissolved oxygen, light, and turbidity. Although the establishment of the nutritional requirements of each life stage was not an objective of this study, satisfactory laboratory diets were defined and verified for each life stage. A comprehensive set of procedures was developed and described in step-by-step manner for use by research personnel wishing to maintain laboratory populations of striped bass for physiological and toxicological use.

This report was submitted in fulfillment of Contract No. 68-03-0316 by the University of Rhode Island under the sponsorship of the U. S. Environmental Protection Agency. This report covers a period from July 1, 1973 to April 15, 1979, and work was completed as of June 1978.

CONTENTS

Foreword. iii
Abstract. iv
Figures vii
Tables. xi
Acknowledgments xv

Section
1. Introduction 1
2. Conclusions. 2
3. Recommendations. 3
4. Development of Current Culture Methods - A Historical Review . . 4
5. Nomenclature, Taxonomy and Morphology. 16
   Nomenclature. 16
   Taxonomy. 16
   Morphology. 22
6. Distribution and Migration 30
   Distribution. 30
   Migration 30
7. Materials and Methods. 41
   General Format. 41
   Source of Material. 42
   General Research Procedures 43
8. Recommended Culture Methods and Bionomics: Embryo 48
   Description of Stage. 48
   Natural Habitat 55
   Environmental Requirements. 55
   Culture Methodology 65
9. Recommended Culture Methods and Bionomics: Larvae 70
   Description of Stage. 70
   Natural Habitat 79
   Environmental Requirements. 79
   Culture Methodology 92
10. Recommended Culture Methods and Bionomics: Juvenile and Subadults. 103
   Description of Stage. 103
   Natural Habitat 106
   Environmental Requirements. 109
   Culture Methodology 124
11. Recommended Culture Methods and Bionomics: Adult. 147
   Description of Stage. 147
   Natural Habitat 152
   Environmental Requirements. 156
   Culture Methodology 162

12. Use in Toxicological Research. 169
Culture Methods Outlined. 169
Previous Studies. 175
13. Population and Stocks. 191
Structure 191
Abundance and Density 195
Natality and Recruitment. 198
Mortality 203
Dynamics of Stock or Population 206
The Population in the Community and the Ecosystem 209
14. Exploitation and Management. 211
Exploitation. 211
Management. 220
15. References 225

FIGURES

Number Page

1 The striped bass, Morone saxatilis (Walbaum). 18

2 Distribution of striped bass along the coast of North America and within freshwater areas of the United States 31

3 Yearly ambient sea water temperatures from flow through laboratory holding tanks during 1974. 44

4 Regional variation in striped bass egg dimensions 49

5 Development of striped bass eggs at 18.8-20°C 52

6 The effect of incubation temperature on the time from fertilization to selected developmental stages before and after hatching. 53

7 Temperature, turbidity, and conductivity measured on the spawning grounds of the Nanticoke River, Maryland, during 1975 57

8 The association of striped bass egg and larval abundance with water temperature in the Hudson River, New York, during 1968. . 58

9 A. Suggested tools for handling eggs and larvae of striped bass so that the animals remain in the water
B. Suggested striped bass egg hatching container modified from a McDonald hatching jar 67

10 Some of the more common abnormalities of striped bass embroys before and just after hatching. 69

11 Developmental stages of striped bass larvae to metamorphosis. . . 71

12 The effect of rearing temperature on the duration of the yolk sac and larval stages of striped bass 74

13 Measurements made on New York 1977 newly hatched striped bass prolarvae after incubation at four temperatures 75

14 Measurements made on New York 1977 striped bass prolarvae at yolk absorption after incubation and maintenance at four temperatures 76

Number Page

15 Percent water content of striped bass prolarvae 78

16 Catch per unit effort of striped bass yolk sac larvae collected by epibenthic sled and tucker trawl at various temperature, conductivity and dissolved oxygen concentrations in the Hudson River, New York during 1975. 80

17 Catch per unit effort of striped bass post yolk sac larvae collected by epibenthic sled and tucker trawl at various temperature, conductivity and dissolved oxygen concentrations in the Hudson River, New York during 1975 81

18 Larval density in the Potomac River, Maryland, during 1974 over the salinity and temperatures reported. 82

19 The effect of delayed feeding on the survival of striped bass stocked at yolk absorption at 24, 21, 18, and 15°C. 87

20 The effect of temperature and delayed feeding on the growth in standard length of striped bass larvae stocked at hatching at 27, 24, 21, 18, and 15°C. 90

21 The effect of temperature and delayed feeding on the growth in dry weight of striped bass larvae stocked at hatching at 27, 24, 21, 18, and 15°C. 91

22 A. Schematic of closed intensive culture facilities used by McIlwain (1975)
B. Basic upflow tank used by Lewis *et al*. (1977) for bass larvae in their recirculating system. 95

23 A comparison of growth rates observed under fixed temperature regimes with those obtained in earlier studies under conditions of increasing temperature 101

24 Abnormalities among striped bass larvae reported by: A - Doroshev (1970) and B - Mansueti (1958) 102

25 Relationship between standard length in millimeters and body weight in grams for post yolk sac larval and juvenile striped bass. 104

26 Relationship between dry weight in grams and wet weight in grams of juvenile and subadult striped bass 105

27 Hudson River sites for juvenile striped bass catch per unit effort at two week intervals beginning at 3/23-4/5 through 12/14-12/27 107

Number Page

28 Abundances of striped bass juveniles (A) and yearlings (B) taken in standard samples during 1973, 1974 and 1975 from the Hudson River. 108

29 Growth in weight and length for juvenile striped bass held in ambient sea water in a cage 115

30 Growth in weight of young-of-the-year striped bass fed at fixed percentages of their live body weight per day on cut squid at ambient sea water temperatures of 18 and 20°C 117

31 Growth in weight of subadult striped bass fed to satiation daily in ambient seawater 121

32 Routine oxygen consumption determined for juvenile and yearling striped bass of wet weight over three temperature ranges. . . . 122

33 Routine oxygen consumption determined for juvenile and yearling striped bass of wet weight before and after feeding over the temperature range of 12 to 14°C and before and after feeding at temperatures of 20 to 22°C 123

34 Ammonia excretion rate determined for juvenile and yearling striped bass of wet weight before and after feeding at 18 to 22°C and before and after feeding at 8 to 12°C. 125

35 Cumulative ammonia excretion by individual juvenile striped bass in seawater and freshwater. Percent survival of all individuals in seawater and freshwater is also shown. 127

36 Holding mortality of juvenile striped bass seined from rivers in Maryland and New York either transported and held in 10^{o}/oo water, or transported and held in freshwater, or transported and held in freshwater until transferred to seawater about August 30th. 131

37 A. Plot of probit estimated time for 50% evacuation for bass of mean wet weight per group for average temperatures indicated
B. Plot of graphically estimated time for 50% evacuation of ration, as a percent of live body weight, consumed by each group 135

38 Growth in weight of juvenile and yearling striped bass fed daily on one of three diets in sea water at average temperatures of 20°C and 16 and 12°C 136

39 Growth in length of young-of-the-year striped bass among 1973, 1974 and 1975 year classes in the Hudson River and mean lengths reported for other coastal populations. 138

Number | Page

40 Composite illustrating lesions and infections of the gill filaments of striped bass observed from histopathologic examination 144

41 Composite illustrating some parasites of striped bass skin and cartilage 145

42 Composite illustrating lesions of the nares and other neoplasma and cysts. 146

43 Fecundity of the striped bass in relation to individual weights . 153

44 Relationship of oxygen consumption to wet weight for striped bass subadults and adults at two temperature ranges 158

45 Schematic of holding facility for adult striped bass. 166

46 Measurements of maximum body depth and standard body length from live (anesthetized) striped bass seined from Maryland rivers (wild) or reared in the laboratory from eggs 170

47 Landing statistics for Massachusetts, Maryland, New York, and North Carolina striped bass fisheries 218

TABLES

Number Page

1 Diagnostic osteological characters of five species of *Morone*. . . 19

2 Distribution of certain meristic characters among striped bass subpopulations. 23

3 Meristic characteristics of striped bass and striped bass hybrids 29

4 Summary of tagging studies involving age 2+ striped bass in the Hudson, Chesapeake, and San Joaquin estuaries 33

5 Summary of the energy content of unfertilized striped bass eggs . 50

6 Relationship between the size of gravid striped bass females and the dry weight of the eggs they produce 51

7 Hatching time of striped bass eggs in relation to water temperatures 54

8 Data on striped bass spawning throughout its range. 56

9 Environmental requirements of striped bass eggs 59

10 Percent survival to hatching of striped bass eggs stocked at various temperature and salinity and temperature, salinity, and dissolved oxygen combinations 61

11 Percent survival to hatching of striped bass eggs exposed to various concentrations of ammonia (NH_3) and nitrate (NO_3) . . . 63

12 The effect of treating filtered river water with penicillin-streptomycin on the percent survival at hatching of striped bass eggs at four temperatures. 64

13 Developmental stages of striped bass, reared at about 17°C, unless otherwise stated, through tranformation 72

14 Average percent composition (carbon and nitrogen) of striped bass prolarvae, larvae at yolk absorption, and fed and starved postlarvae reared at four temperatures. 77

Number Page

15 Environmental requirements of larval striped bass 83

16 Percent survival of striped bass prolarvae after 48 hours exposure to various ammonia (NH_3) concentrations, temperatures, salinities and pH's 85

17 Caloric and percent composition of some live larval food items. . 98

18 Instantaneous growth coefficients for delayed feeding groups at five constant temperatures. 100

19 Environmental requirements of striped bass juveniles and subadults 110

20 Preferred foods of striped bass 113

21 Summary of growth data for each group of striped bass fed one of two diets at 10 and 20°C. 118

22 Absorption and conversion efficiencies calculated for striped bass fed one of two diets at 20°C and compared to growth efficiency from Table 21. 119

23 Excretion rates measured forth-eight hours after last meal for individual juvenile and subadult striped bass 126

24 Parasites and diseases of striped bass reported from the literature and this study 128

25 Calories and percent composition of some foods of juveniles and subadult striped bass 134

26 Treatment recommended for some of the parasite groups common to striped bass. 141

27 Incidence of lesion type. 142

28 Age and size at first maturity for striped bass 148

29 Length-weight relationship for striped bass 149

30 Comparison of growth of striped bass from various areas 150

31 Relationship of gonad weight, egg number, body length and body weight among striped bass of various ages captured in a number of areas 151

32 Percent survival through hatching of striped bass eggs from artificial and natural spawnings. 154

Number Page

33 Environmental requirements of striped bass adults 157

34 A. Daily food consumption levels for striped bass fed to satiation at different ambient sea temperatures
B. Evacuation rates for striped bass fed daily to satiation. . . 159

35 Excretion rates typical for adult striped bass at two temperatures at 30°/oo 160

36 Growth rates typical for adults maintained in seawater and fed daily 167

37 Summary of optimal rearing conditions for the various striped bass life stages. 173

38 Toxicity of substances to striped bass larvae 176

39 Toxicity of substances to juvenile striped bass 177

40 Testing conditions for striped bass bioassays 179

41 Water quality of bioassays using striped bass 180

42 Analysis of chemical substances used in striped bass bioassays. . 181

43 Residue concentrations of heavy metals reported in muscle (flesh) tissue from wild striped bass 187

44 Summary of hydrocarbon residues reported in muscle (filet) of striped bass. 188

45 Summary of hydrocarbon residues reported in striped bass ovaries 189

46 Examples of annual densities of striped bass reported for different areas 197

47 Estimates of egg and larval production and survival rates 200

48 Estimates of survival and mortality rates for some striped bass stocks. 204

49 Summary of information available on striped bass fisheries. . . . 212

50 Percentages of striped bass landed caught by each gear, by state, for 1962-66 and 1967-71. 213

51 State regulations governing striped bass fishing. 214

Number Page

52 Estimates of striped bass fishing efforts from survey of anglers 216

53 Striped bass landings by marine state in the United States in metric tons. 219

ACKNOWLEDGMENTS

The authors wish to thank the many individuals and groups who assisted us in many aspects of the work during the course of this study. We wish to thank particularly the laboratory assistants who served for various periods of time during the course of the investigation: John Hartley, Michele Cyr, Norman Kahn, Aimee Keller, Linn Meller and Steve Clements. We wish to acknowledge and thank the following individuals and groups who assisted us in collections of various life stages: Jack Bayless, Moncks Corner striped bass hatchery, S.C.; Joe Boone, Maryland Fisheries Administration; Halleck Fish Co., Shady Side, Md.; Tallman and Mack Fish Trap Co., Newport, R.I.; and Consolidated Edison Co. of New York, the owners of the Verplank, N.Y. experimental striped bass hatchery operated by personnel of Texas Instruments, Inc. All of the figures were prepared by the Graphics Department of the Graduate School of Oceanography, University of Rhode Island, whom we thank for their perseverance. Drs. Richard Wolke and Peter Belinsky of the URI Marine Pathology Laboratory performed numerous histopathological examinations of striped bass specimens and provided valuable diagnostic advice. Special thanks are offered to Susan Proulx and Janice Steele for their patience and ability in typing the text and tables through many drafts. Deborah van Dam provided important editorial comments. Finally we wish to thank Alan Beck, our project officer, for his patience and encouragement through out this study.

SECTION 1

INTRODUCTION

Striped bass, Morone saxatilis, is an important commercial and sport fish species with a center of distribution between the Hudson River and the mouth of Chesapeake Bay. Individuals of this species ascend major rivers to spawn, use coastal estuaries as nursery grounds, and as adults make seasonal migrations along the coast rarely straying more than five miles from the shoreline. Because it passes its entire life cycle in the waters immediately adjacent to the Boston-Washington,D.C., megalopolis, it is subjected to the most intense effects of man-made pollution and environmental alteration. In spite of these abuses, the Atlantic population of striped bass has until recently enjoyed great abundance. Although in the past considerable amount of research has been done on the culture of the species for stocking into southern reservoirs, no reliable culture methodology has been developed for maintaining all of the life stages of the striped bass in the laboratory where the effects of various pollutants may be determined in physiological studies and bioassay experiments.

The present study was undertaken to develop a reliable culture protocol for all life stages of the striped bass. Armed with such a protocol, researchers will be in a better position to examine the effects of water borne pollutants on this resilient but vulnerable species.

SECTION 2

CONCLUSIONS

During the course of this study all of the life stages of the striped bass from egg to adult were successfully maintained under laboratory conditions. The temperature, salinity, dissolved oxygen, light and turbidity requirements of all life stages were either determined empirically, approximated from environmental data, or where reported by other workers corrobrated in our laboratory. Optimum and survival limits for each of these parameters were, where appropriate, specified. By maintaining conditions within these bounds, striped bass eggs were repeatedly reared through to the juvenile stage. A population of striped bass adults were successfully maintained in captivity for five years. Despite repeated attempts, we were unable during the course of this study to successfully induce spawning in the laboratory. Sexually mature adults of both sexes, however, did occur among our captive population. A step-by-step culture methodology has been prepared for use by future workers.

SECTION 3

RECOMMENDATIONS

Although striped bass reach sexual maturity from two to nine years after hatching, it is, we believe, possible and desirable to perform life cycle studies using this species. In this study we were unable to close the circle and demonstrate an egg-to-egg culture capability. Continued work along this line would be highly desirable. The culture requirements of larval and juvenile striped bass as well as adults may be easily met in the laboratory. We urge more frequent use of this important species as a subject for laboratory investigators. Increased knowledge of the effects of food and/or water borne contaminants of the various life stages of the striped bass could and should be used as a basis for efforts to diagnosis and remedy the recent dramatic decline in the recruitment of the species into the sport and commercial fisheries on both the East and West Coasts.

SECTION 4

DEVELOPMENT OF CURRENT CULTURE METHODS - A HISTORICAL REVIEW

Striped bass culture had its beginnings in the latter half of the nineteenth century during the childhood of American fish culture. Commercial and subsistence fishing had been an important part of the North American economy since the first settlers arrived. As early as the mid-1700's many New England fisheries had been completely eradicated through the combined effects of virtually unrestricted fishing effort, dam building, imprudent agricultural practices and river pollution. By the mid-nineteenth century many coastal, estuarine and inland fisheries were in a state of decline. In addition, other fisheries, while apparently not suffering directly from overexploitation, were affected by fluctuations in abundance which caused economic dislocations in the fishing industry.

The techniques of salmonid culture developed and described in Europe by Stephan Ludwig Jacobi in 1764, were rediscovered and popularized in France by Joseph Remy and Antoine Gehin in the late 1840's. In his first report, M. Coste, then director of the first fish hatchery to be built by the French government, in 1852 stated "There is no branch of industry or husbandry, which with less chance of loss, offers an easier certainty of profit." (Davis, 1967, p. 6). This ebullience was to characterize the hatchery movement in Europe and in North America for the next half century.

In 1853 Theodatus Garlick, of Cleveland, Ohio, was the first American to attempt and succeed at fish culture. His pioneering experiments with brook trout paved the way for the entrepreneur-culturists, Seth Green, Livingston Stone, and Thaddeus Norris, the acknowledged fathers of American fish culture. By the late 1860's, 19 states maintained hatchery operations as did numerous private culturists. During this period it was generally realized that the stocking of artificially propagated fish was a more politically acceptable palliative for the problem of overfishing than any efforts to limit the catch (Bowen, 1970). However, there was little incentive to stock interstate waters.

In 1868 the shad fishery was in a poor state in many areas. In that year the federal Commissioner of Agriculture was petitioned by culturist Seth Green and several others to sponsor efforts to propagate shad. By 1872, the U.S. Fish Commission, which was formed a year earlier, under the leadership of its first Commissioner, Spencer F. Baird, received the mandate from Congress "for the introduction of shad into the waters of the Gulf States, of the Mississippi Valley and of the Pacific States, and of salmon, whitefish, and other useful foodfish in the waters of the United States to which they are best adapted..." (Bowen, 1970, p. 82-83). Baird, working with and around the recently formed American Fish Cultural Society, the precursor to the American Fisheries Society, led the federal effort to restore depleted fisheries, both inland and coastal, through artificial propagation. The shad restoration effort was the heart of the federal fish culture program during the early years of the Fish Commission's activities in this area.

Marcellus G. Holton, who was employed by the fish commission to undertake shad spawning operations on the Roanoke River in North Carolina, reported in 1874 to Commissioner Baird that he had successfully spawned and hatched striped bass, or rockfish, in May of 1873. His was the earliest report of attempts to propagate this species. E. H. Walke, also of the Fish Commission, reported successful efforts at spawning and hatching striped bass eggs in 1879, again incidental to shad spawning operations. S. G. Worth of the North Carolina Sub-department of Fish and Fisheries described details of his successful attempts to spawn and hatch striped bass taken in Albemarle Sound in the spring of 1880. He concluded: "...it may be inferred that rock-fish eggs are as easily fertilized as those of shad, and it would in addition appear that a less amount of milt is necessary. It would further appear that they are more hardy, even admitting large amounts of sand and other mechanical substances into the water while undergoing impregnation. ...it occurs that it only remains to ascertain the spawning localities of the parent fish when their propagation will follow." (1882, p. 176). Worth appeared to be the first culturist to undertake efforts specifically for the purpose of hatching striped bass. In 1884 he reported: "...I established at Weldon (North Carolina), quite late in the season of 1883, an exceedingly crude establishment, containing sixty-five McDonald jars, equipped as if for very crude shad or whitefish hatching. The station was provided with a force of five experts, a force rather too small, though efficient." (1884b, p. 210-211). Worth and his force succeeded in hatching and releasing 50,000 striped bass fry from the estimated 1,000,000 eggs they had taken. The only difficulties he mentioned were the delicacy of egg chorions late in development and the lack of fine enough screens to retain the newly hatched larve. He concluded that "...there seems scarcely a doubt of securing a great supply of eggs, thus opening a means of propagating the choice, valuable striped bass." (1884b, p. 212). Thus it was with great optimism that the first century of striped bass culture was begun.

Since efforts to hatch striped bass were offshoots of shad culture operations, no special equipment or techniques were used in these early culture experiments. At times eligible males and females of either species, striped bass or shad, were unavailable, prompting these early investigators to try and cross-fertilize the two species. Whether or not these efforts were undertaken with the serious expectation of success is unknown. Worth (1882) fertilized striped bass eggs with shad milt and observed 5-6% survival through hatching, with some larvae surviving for an additional 12 days. Writing in 1887, Ryder noted "It is rather extraordinary that the striped bass should so readily lend itself to the purpose of cross-fertilization with other closely allied species, such as the white and yellow perch, but is still more astonishing that it should be possible to cross this species with another belonging not simply to a different family, but even to a widely different order and sub-class." (p. 524). Ryder then described what he felt was incontestable evidence that reciprocal crosses were possible between shad and striped bass. In addition, he quoted a publication by R. B. Roosevelt of New York in which he too reported a viable cross between shad and bass. Ryder's report was the last to mention inter-ordinal crosses involving striped bass. Hybridization between the striped bass and its congeners, however, was to receive much additional attention eighty years later (see Section 5).

It was within the spirit of the fish culture movement of the period that efforts were made to establish striped bass in California waters. Shad fry had been transported to California in 1871 two years after the completion of the transcontinental railroad. By 1880 the Atlantic shad had been established from San Francisco Bay to Vancouver. No doubt heady with the success of the shad introduction, S. R. Throckmorton, the chairman of the California Fish Commission, engaged Livingston Stone to import to the west coast young striped bass, lobsters and several other Atlantic coast species. Striped bass were planned to be included among the species transported west by Stone in 1874 but for logistic reasons were not. However in 1879 he did succeed in transporting 133 juvenile striped bass captured in the Navesink River, New Jersey, to San Francisco Bay where they were stocked in Carquinez Strait between the fresh and salt water sections of the Bay. The initial stocking was apparently an instant success. Eleven months after the fish were stocked a 12 inch specimen was caught. By the time a second planting of approximately 300 juveniles was undertaken by J. G. Woodbury in 1882 striped bass appeared to be well established (Shebley, 1927).

Spectacular successes such as the striped bass introduction in California gave credence to the proponents of artificial propagation as a fisheries management tool. Successful transplantations such as this, however, did not allow the culturist an opportunity to perfect the techniques of spawn taking and egg hatching that were required in other fisheries. Hatching techniques used in early experiments with striped bass were essentially the same as those used for shad. Worth (1882) noted that striped bass eggs were larger and somewhat more bouyant than those of shad. He also observed that striped bass eggs did not require the same water volume as shad eggs when McDonald jars were used. This observation no doubt came about as a result of attempts to use flows equivalent to those used on shad, which because of the lower density of bass eggs would have resulted in washing the eggs out of the hatching jar or rapid clogging of aquarium screens, where these were used. Worth apparently also hatched eggs in fabric cones in floating live cars, a technique also used for hatching shad. Brice, in his 1898 Manual of Fish Culture, observed that "The tidal apparatus, such as is used for cod and tautog eggs, is adapted to hatching the eggs of this fish" (p. 185). Brice does not mention whether or not the 'McDonald tidal egg hatching box' was ever, in fact, used to hatch striped bass eggs. The actual hatching of fertile eggs never appears to have been an important problem to these early culturists.

From the very beginning, however, finding female striped bass in spawnable condition was a problem. Even initial enthusiastic commentary on Holton's 1874 announcement that he had successfully spawned striped bass was hedged with the proviso that "If localities can be found where rockfish may be taken in sufficient numbers in the breeding season, the increase of this species is probably as sure to be as effected as that of the shad has been." (p. 554). In 1882 Seth Green observed to the members of the American Fish Cultural Association: "There have been but a few sturgeon and striped bass hatched artificially. The reason that there have not been more is that it is so difficult to get the mature fish when the spawn is ripe." (p. 37). He then proposed holding potential spawners in live cars until they matured. In his 1884 report to the American Fish Cultural Association, Worth noted that "It is not known at what points ripe fish of this species can be found in greatest abundance, but in our present state of knowledge, Weldon, North Carolina, presents the greatest number." (p. 209). Of Weldon he noted:

> "Although large quantities of striped bass are taken during the several months by the large seines and pound nets seaward, there appears to be no one point where the eggs in a condition proper for fecundation can be found so abundantly. At the particular point named, the fall is so great that ordinarily, owing to a lack of a great volume of water to smooth over the falls, the fish are unable to pass directly over, and in consequence are detained at the foot of the falls." (1884b, p. 209).

While realizing that the Weldon site was unique, Worth felt that there were other suitable spawn taking areas downstream in the Roanoke "...with the system of impounding, there seems scarcely a doubt of securing a great supply of eggs," (p. 212). Worth's prediction proved to be somewhat optimistic. Difficulties in obtaining ripe striped bass were encountered elsewhere as well. Fish Commission culturists at the Havre de Grace station, Chesapeake Bay, were not successful in artificially fertilizing and hatching striped bass because of difficulties encountered in trying to obtain ripe males and females at the same time. The construction of live holding facilities was suggested but apparently none were built. Norny (1882) suggested the use of an enclosed pond near the spawning grounds on the Delaware River as a means of procuring ripe females. Although he demonstrated that holding females was possible, no major cultural effort ensued. The Weldon station was operated by the U.S. Fish Commission and later the U.S. Bureau of Fisheries well into the Twentieth Century (U.S. Fish and Wildlife Service, 1904-). In 1913, Snyder reported on some of the improvements in hatching operations that had taken place at Weldon over the years. Even at Weldon the problem of obtaining females in the proper condition for spawning persisted. Commission culturists obtained their ripe females from commercial fishermen working along the river. Snyder noted "...during the past four seasons I... have taken the eggs from only five fish, which were all the ripe fish I saw caught. ...Has it been proven that these fish will not ripen in crates?" (1913, p. 96). In 1915, Snyder reported that in fact they would ripen in live cars. Although he was not pleased with the construction of the live car he used, Snyder was able to conclude after his experiences during the spring of 1914 that nearly ripe striped bass ripened in confinement and that some of the eggs of those fish which ripen in confinement produced good results. In all, seven of the 30 fish penned that spring in Weldon spawned. Two of these "cast their eggs" in the car between examinations. In recapitulating the results of his penning experiments Snyder observed that the only females yielding eggs were those with very soft abdomens at the time of capture. Among these promising individuals there was still great variation in the degree of survival that was realized. What success he did achieve he attributed to a large extent to the great care he exercised in handling his brood fish. Snyder's spawners were captured on a 'slide' of a wooden wier in the river; as a result there was little capture damage to the fish.

Based on observations by fishermen of large concentrations of ripe females in the area during the years 1903 through 1905, the California Fish and Game Commission decided to locate a hatchery near Bouldin Island on the San Joaquin River (Scofield, 1910). The hatchery began operations in 1907. Fishermen brought in ripe females to hatchery personnel who stripped them, applied milt and transferred the fertilized eggs to McDonald hatching jars. During the first season the hatchery was filled to capacity. Survival among the lots of eggs received ranged from over 50-60% to about 5%. The range in hatching success was attributed to water quality and defects in the hatchery methods used. The hatching rate among successful spawns was higher than had been reported for hatching operations on the Atlantic Coast. During the following season the expected run of spawners failed to materialize. Among the fish that were examined it was found that spawners exhibiting low rates of survival also showed a low rate of fertilization based on microscopic examination of eggs during the first few hours of development. Using the microscope, each lot of eggs was examined for the percent undergoing normal cleavage. Variations in survival which had been laid to handling and water quality were now attributed to differences in the degree of ripeness of the females spawned. Immature females were observed to have lighter colored eggs than the dark bottle-green eggs of fully ripe individuals. Filamentous fungus developed on dead eggs in the hatching jars. It was controlled using a 1:100,000 solution of copper sulphate with no apparent ill effects. This was the first recorded instance of the use of chemical treatments in striped bass culture. Both wet and dry methods of fertilization were tried. The wet method yielded only slightly better results. Having observed that the majority of the fish taken had not reached the necessary stage of ripeness for successful egg taking, Bouldin Island culturists constructed a large holding pen, although reports from the Atlantic coast indicated that striped bass were difficult to maintain alive in pens and that past efforts to hold females until they reached ripeness had not met with great success. During the 1909 season the run of bass was as poor as it had been the year before; however, 50 females were caught and penned. Most of the fish taken using gill nets died within a few days. Males and immature females survived longer than ripe females which lasted no more than 24 hours after capture. It was concluded that penning brood fish was practical only where all forms of handling could be kept to a minimum. Few eggs were taken from all of the fish captured in 1909. The following year the run improved, but all of the females taken were immature. Ripe males were plentiful. The Bouldin Island hatchery was abandoned after the disappointing results during the 1909 and 1910 seasons. Although ultimately unsuccessful, the Bouldin Island operation was the first to bring to bear up-to-date biological techniques in the examination of spawning adults, eggs, and larvae. The observations reported by Scofield and Coleman (1910) during their biological experiments at Bouldin Island were the first systematic investigations of some of the biological problems encountered in spawning and hatching striped bass.

The experiments at the Bouldin Island hatchery in California and Snyder's (1915) efforts on the Roanoke River, N.C., a few years later both pointed out the major problem in striped bass culture to date, namely, the extreme variability in the degree of ripeness that occurs even among females that are on the spawning grounds and are nearly ready to spawn. It was clear that, as Scofield pointed out, "The taking of a female bass with ripe eggs was evidently a lucky chance,..." (1910, p. 106) and that holding females until they ripened, although it increased the odds of finding one which was in precisely the right stage of ripeness, was at best a difficult proposition.

Early culturists looked upon propagation as a means of helping nature produce fish fry. In most instances newly hatched larvae were stocked directly into the waters from which their parents were captured. When distant waters were to be stocked fry were transported in shipping cans with no particular care being given other than to shield them from temperature extremes and provide periodic water changes. Little attention was given to the cultural requirements of the larvae or later developmental stages. In his 1884a report Worth stated: "As far as the keeping of the fry is concerned there is no difficulty; in former experiments I have found no difficulty whatever in keeping them alive in ordinary shipping cans a period of twelve days with moderate changes of water through the tin strainer tube." (p. 228). In 1904 Worth reported that he had reared an unspecified number of larvae for four weeks in a "crudely constructed pool near the hatchery door." He states: "I do not think that partial rearing in ponds could be other than successful, as the water in the temporary pool at Weldon was of very high temperature and almost stagnant." (p. 226).

The hatchery at Weldon has operated with only minor interruptions since Worth's time under the administration of the Bureau of Fisheries (and its successors) and the state of North Carolina. Raney writing in 1952 noted: "Experience at the Weldon hatchery has shown that the fry are not held successfully for more than 12 to 24 hours after hatching without high mortality, ... fry must be handled very carefully to avoid large losses, and the longest haul successfully accomplished took about two hours." (p. 44). Although Raney suggested that crowding and polluted water may have been responsible for this extreme tenderness among striped bass larvae spawned at Weldon, the difference between the problems he cite and the minor difficulties evidently encountered by Worth are noteworthy. The techniques employed at the Weldon hatchery underwent very little change between 1906 and the late 1950's. Pearson (1938) mentioned successful experiments undertaken in 1937 at Weldon and Edenton, North Carolina, aimed at rearing larval striped bass in aquaria and outdoor ponds through the introduction of natural foods such as Daphnia. However, he did not give details of this work.

In 1942 the South Carolina Public Service Authority completed a hydroelectric project which involved the damming of the Cooper and Santee

Rivers on the coastal plain of South Carolina. Prior to the completion of the project minor runs of striped bass occurred in the Santee River with none of any importance in the Cooper River. The damming effort diverted all river flow from the Santee River eliminating the run on that stream and channelling it through a hydroelectric dam equipped with a navigation lock which was connected by a tailrace canal to the Cooper River. The resulting impoundments, Lakes Marion and Moultrie, covered an area of 160,500 acres with a total shoreline of 415 miles. Shortly after the impoundment had been completed, sportsmen's catches of striped bass upstream of the dams increased markedly. In addition, the increased flow in the Cooper River-Tailrace Canal System provided conditions which fostered an increased seasonal run of striped bass below the hydroelectric dam. Although no effort was made to encourage the passage of anadromous species through the navigation lock into Lakes Marion and Moultrie, catches of menhaden, alewives, and American shad in the impoundments suggested that the dams were not an impenetrable barrier to normally migratory species. Observations of large numbers of juvenile striped bass in the two lakes suggested that some reproduction might be taking place above the dams. In 1955 Scruggs and Fuller reported the occurrence of striped bass eggs and larvae well above the dams at the mouths of the Congaree and Wateree Rivers, strongly suggesting that striped bass were capable of spawning in an entirely landlocked situation, a previously unsuspected case. Surber (1958) in a review of the results of various attempts to introduce adult striped bass into fresh water impoundments both before and after Scruggs and Fuller's discovery noted that, on the whole, these efforts had met with meager success. Only in the case of the Kerr Reservoir, a large impoundment on the Virginia-North Carolina border, was there any evidence of successful reproduction. Although striped bass, introduced as juveniles or adults often thrived under landlocked conditions offering sportfishing opportunities and the promise of controlling exploding populations of undesirable species such as gizzard shad (Dorosoma spp.), it was concluded that all but a very few impoundments failed to supply fast flowing tributary streams which were felt to be necessary for successful spawning and egg survival. Stevens (1965) opined that "The spawning recruitment dictates that unless the freely-spawned striped bass eggs remain suspended in a current until hatching, they will settle to the bottom and suffocate." (p. 526). Reasoning that "the reservoirs of South Carolina, other than Santee-Cooper, are physically deficient as to the spawning requirements of the striped bass and that reproduction is doomed for this reason. ... , a hatchery was constructed in 1961 at Moncks Corner in order to circumvent this limiting factor to the successful establishment of striped bass throughout the state." (p. 526).

The Moncks Corner Hatchery, the first to be established since the closure of the Bouldin Island hatchery in 1910, was inspired in its configuration by the Weldon hatchery which had by that time been operating more or less continually for over 70 years. Weldon was located below Weldon Rapids which formed an impasse to upstream migrants on the Roanoke River. The Moncks Corner operation was located on the tailrace of the Pinopolis Dam at the head of the Cooper River-Tailrace Canal System. Here each spring striped bass in spawning condition were known to congregate in great numbers. It was felt that a situation like that at Weldon would be created. Hatchery personnel rather than commercial fishermen were to capture potential spawners in deference to South Carolina law which proscribed all commercial fishing for striped bass and prohibited all fishing in a sanctuary area just below the dam. Of 900 fish examined in 1961 none were found to be fully ripe. While many fish were concentrated just below the dam, it appeared that actual spawning took place over a stretch of several miles below the dam. Concluding that the time between ovulation and the actual release of eggs must be very short, it was decided to abandon attempts to capture brood fish in precisely the proper condition for artificial spawning.

Pickford and Atz's (1957) review, The Physiology of the Pituitary Gland of Fishes, and several succeeding studies by personnel of the U.S. Fish and Wildlife Service, had demonstrated the efficacy of hormone injections as a means to induce precocious spawning in a number of fish species maintained in captivity. Between 1962 and 1965 a series of experiments were run at Moncks Corner under the leadership of Robert Stevens of the South Carolina Wildlife Resources Department, which led to the development of a procedure which could be used for the routine induction of spawning in Cooper River females. This provided a basis for an extremely productive fry production program at Moncks Corner. Stevens (1966) investigated a number of hormone injection protocols before arriving at an optimal means of assessing maturity, determining the proper timing and dosages of hormone and learning to predict with great precision the time at which brood fish should be stripped to ensure maximum egg survival. In addition to the use of human chorionic gonadotropin, which was ultimately adopted as the hormone of choice for this work, he investigated the use of a variety of other hormone preparations. These included follicle stimulating hormone, pituitary luteinizing hormone, thyroid stimulating hormone, estrogen preparations, testosterone, and fish pituitary glands. Only chorionic gonadotropin and follicle stimulating hormone induced ovulation in striped bass females when used alone. Other procedural improvements effected during this period included the construction and use of holding ponds, the use of AC electrofishing techniques to capture broodfish, and of constant temperature well water to supply the holding tanks and hatch house. McDonald jars were used in the hatchery in much the same configuration as they had always been used, but some of the troublesome aspects of their use such as air entrainment and pressure variations were circumvented. Two phenomena discovered by Stevens and his co-workers proved to be a great help in explaining why so many earlier attempts at holding fish until they were ripe had failed. 'Over-ripeness ," the retention of already ovulated eggs in the ovary without releasing them, led to the production of eggs with very low fertilizability. It was found that there was a period of not over one hour between ovulation and the onset of over-ripeness. Eggs not stripped in this period showed very low survival through hatching. Over-ripeness was avoided through the periodic inspection of egg samples removed from the oviduct using a catheter tube. Abortion, or the ovulation of immature eggs, was observed in some cases but the regular use of hormones and regular inspection prevented abortion from being a serious problem. There was evidence, however, that hormone induced ovulation increased the number of immature eggs that were released. These eggs could be fertilized but seldom survived through hatching. Earlier workers at Bouldin Island had apparently tried to strip a number of fish with immature or golden colored eggs.

The hormone techniques developed at Moncks Corner in the early sixties were very effective in the hands of those practiced in their use. Experienced culturists became expert at estimating the time to strip females based on relatively few inspections. Minimizing handling reduced the likelihood that females would die before viable eggs could be removed. The use of the anesthetic MS-222, applied topically to the gills prior to stripping increased the number of females which survived the spawn taking process.

The use of the techniques developed at Moncks Corner resulted in the production of eggs which survived well through hatching. Estimated percent hatching increased from an average of 7.3% in 1962 to 31.0% in 1964 when 100,000,000 fry were produced (Stevens, 1966). Fry produced during this period were held in aerated aquaria for up to three days after hatching at which time they were stocked into grow-out ponds. As Stevens in South Carolina was perfecting spawning techniques which were leading to greatly increased levels of fry production, work was proceeding at the Edenton National Fish Hatchery, at Edenton, North Carolina, aimed at growing striped bass fry to fingerling size.

Newly hatched fry produced at Weldon had been used to stock a number of inland areas in the south. Non-reproducing populations had been established in several cases. Owing to the apparent lack of suitable spawning habitat in many inland water bodies, the only way in which striped bass could be maintained in these lakes to support sportfishing and rough fish control was through continued introductions of hatchery reared fry from Weldon or Moncks Corner. 'Put, grow and take' management of many lakes and reservoirs involved the production of predigious numbers of fry each year. Stevens noted "On a put, grow and take basis, however, low survival could not be tolerated because the inherent inefficiency would make the concept economically unjustifiable." (1967, p. 2). As of 1965 the Kerr Reservoir was the only inland water to have a population of striped bass which had been created by stocking fry. Stevens (1967), however, cited information which cast doubt on the validity of even this claim. A total of three million fry stocked between 1952 and 1954 from Weldon were claimed by some to be the basis for the population of adults that appeared later. It was reasoned that with the low survival among fry stocked shortly after hatching a protracted yearly stocking program would be needed to yield any significant results. The need for better survival was evident. Anderson (1966) reported on efforts made in 1964 at the Edenton Hatchery. Lots of fry obtained from Weldon were divided between hatchery troughs and fertilized ponds. Those maintained in troughs on a diet of emulsified shrimp ultimately died, although they did appear at first to be consuming the diet provided. Fry stocks in the pond, which had been limed and fertilized with soybean meal and applications of 20-20-10 inorganic fertilizer, survived on the micro-crustacean populations induced by heavy fertilization and grew rapidly. As they grew larger, natural food was supplemented with ground herring and trout food. The pond which had been stocked May 20 was seined on August 11 and 30,000 fingerlings recovered with an average weight of 4.5 grams each. In 1965, Sandoz and Johnston repeated Anderson's success in rearing striped bass to fingerling size in ponds in which the zooplankton population had been enhanced by heavy fertilization. Like Anderson they had poor luck in attempts to maintain larvae on non-living food. Larvae were, however, reared to fingerling size on zooplankton netted from the hatchery pond and fed to fish in aquaria.

Once it had been shown that the production of pond reared fingerlings was a practical proposition, a number of fish and game agencies in the southeast and mid-west became involved. Reviewing the results of pond rearing experiments carried on in over a dozen different state agencies, Stevens (1967) concluded as follows on larval feeding:

> "If possible, fry probably should not be stocked when younger than four to six days old because they have a tendency to settle to the bottom where suffocation may occur.
>
> A four-day-old fry, on the other hand, is continuously in motion and by the eighth day, starts feeding.
>
> There seems to be no doubt that zooplankton is essential to the life of striped bass fry from day eight until a yet to be determined time, perhaps until the post larvae reaches at least one inch in length. After this time, supplemental feeding alone may be sufficient." (p. 4)

Although most practicioners were able to obtain satisfactory zooplankton blooms in their rearing ponds, they often found it difficult to maintain a sufficient zooplankton density over a long period. A variety of supplemental feeds were used by the various groups, including trout food, ground meat and fish, and live fish. Fingerlings consumed a variety of these feeds but, as always, it was difficult to determine to what extent the fish relied on the food offered and to what extent they lived off the planktonic and benthic populations of the ponds in which they were kept.

In 1975, Braschler summarized the development of pond culture techniques. Bonn et al. (1976) provided an extensive section on pond culture. They included pond preparation (liming and fertilizing), pre-stocking checks (zooplankton abundance, predators, and temperature), and stocking. The suggested optimum stocking rate was100,000 fry/acre. Plankton, vegetation and insect control during growth period (4 to 6 weeks) prior to harvest as fingerlings werealso discussed. Powell (1976) described brackish water culture in Alabama. Rees (1974) suggested that further investigations into rearing to advanced size in raceways should be explored. This would allow closer observation of feeding, growth, mortality, and diseases than is possible in ponds.

During the later part of the 1960's and early seventies the Edenton National Fish Hatchery made additional strides in perfecting the extensive rearing of striped bass fingerlings (Meshaw, 1969; Bowker et al., 1969; Ray and Wirtanen, 1970; Wirtanen and Ray, 1971). Fry which had developed functional mouth parts were routinely stocked in rearing ponds and were recovered as fingerlings several months later. Stocking was performed in

April and May when newly spawned fry were available. Fingerlings were recovered early in the summer and transported to their assigned stocking sites. During the years when these systematic investigations were underway a number of new procedures developed and, concurrently, new problem areas were revealed. Since the aim of most of these studies was to improve production rather than to perform basic research, investigators were unable to follow up on all of the observations they made. It was found that larvae and juveniles under 8 weeks of age fed exclusively on planktonic food and made virtually no use of benthic organisms in the Edenton ponds. Efforts were also made to determine to what extent supplementary feeding was necessary in pond raised striped bass fry. Results obtained during the 1967 and 1968 seasons revealed that fry receiving trout food or ground herring as a supplemental ration had a lower survival rate than those in ponds in which zooplankton alone was present. Where zooplankton was abundant the juvenile bass grew faster and enjoyed better survival when no supplemental food was supplied. Although the fish were observed to eat the supplementary ration when live food was abundant, they apparently showed little inclination to do so. It was felt that if young fingerlings were to be weaned onto complete reliance on the ration they were being supplied that they would have to be confined and taught to eat the supplemental food. Advanced fingerlings showed better growth and survival when supplied with ground herring than when offered only dry trout pellets. Groups which underwent an abrupt change in ration showed the lowest survival and growth. Palatability was apparently an important factor.

In a later series of experiments dry diets alone were used, dispensed by hand and through the use of automatic feeders. Lots fed from automatic feeders showed a slightly better feed conversion, however it was thought that this could be due to the more frequent feedings that the feeders made possible.

The methods used at the present time in most hatcheries involve feeding brine shrimp during the pre-pond stocking phase, especially if this extends beyond yolk sac absorption. After stocking fry into ponds, supplemental feeding may be desired either to augment the zooplankton supply or to increase production. Bonn _et al_. (1976) suggest feeding artificial trout feed at the rate of 5 pounds per acre per day after bass are three weeks of age. This rate can be increased to 20 pounds per acre per day at harvest time. Harper and Jarman (1972) found supplemental feeding of fry and fingerlings in ponds increased production, albeit identification of these commercial diets was lacking in the food habit studies conducted concurrently.

It became clear that in handling fingerlings, both during restocking and during harvest, great care was needed to prevent undue stress to the fish. Pond harvesting techniques were described by Bonn _et al_. (1976). These included the use of glass V-trap to harvest 80% of pond crop from properly constructed ponds, catch basins, seines with partial draining, or using nursery ponds built to stock directly into lakes. Transport of fingerlings harvested from ponds to stocking destination was also described by Bonn _et al_. (1976). They suggested holding for 24 hours and treating with Furacin at 100 to 500 ppm with 1% salt for 2 to 5 hour periods prior to shipping. Fingerlings were then hauled in tank trucks with 1% salt (NaCl) and 21 ppm MS-222 or 0.35 ppm Quinaldine to reduce activity. Acriflavine at rate of 1 ppm could also be used during hauling. Transport densities suggested were 1/4 to 1/2 pound per gallon with good aeration.

The problem of handling gravid adults was addressed during early studies, but never resolved. Adult females which were being held for spawning purposes frequently developed the 'red-tail syndrome'. This condition started as a hemorragic area on the caudal fin. The reddened area generally spread over the caudal penducle and sometimes over the entire posterior portion of the body. Most often the affected individual became unresponsive and died. Both Edenton workers and Stevens in South Carolina tried a number of antibiotic and antiinflamatory drugs with no success. Frequent pre-ovulation mortalities among hormone injected females was also a problem that was never explained or resolved. Losses among brood fish prevented their repeated use in successive seasons. At the present time, hatchery procedures for broodfish transport throughout the south eastern states generally follow those outlined above for fingerling transportation.

Other pathologic conditions observed among captive striped bass included columnaris disease, and hemorragic gill disease. Posthodiplostomum trichodina was a problem external parasite. Losses attributable to bacterial diseases were most often associated with crowded conditions, either in ponds, tanks or raceways. Disease and parasite problems encountered during hatchery and pond production of striped bass have been described by Bowker et al. (1969), Regan et al. (1968), Hughes (1975), Hawke (1976), and Bonn et al. (1976).

Today, striped bass reared in ponds are used primarily for stocking of lakes, reservoirs and impoundment areas for sport fishing and shad (Dorosoma spp.) control. Most of this production is carried out by federal and state hatcheries and agencies. The states producing striped bass include Alabama, Arkansas, Florida, Georgia, Kansas, Kentucky, Louisiana, Mississippi, Missouri, Nebraska, North Carolina, Oklahoma, South Carolina, Tennessee, Texas, and Virginia. Production and survival by agencies of these states for 1972-1975 was tabulated in Texas Instruments (1977c), where the grand total production for these years combined exceeded 14,000,000 fingerlings. Production by federal hatcheries for the same years was also tabulated in Texas Instruments (1977c). The total fingerlings produced from these hatcheries was in excess of 15,000,000. Production data for this and other species by federal hatcheries is available from the U.S. Fish and Wildlife Service (1904-). Federal hatcheries producing striped bass for stocking are in Alabama, Arkansas, Florida, Georgia, Kentucky, Louisiana, Mississippi, Missouri, North Carolina, Oklahoma, and South Carolina.

Stocking of underutilized brackish water nursery grounds was investigated in Virginia by Merriner and Hoagman (1972). Striped bass have been reared in the Hudson River for population and entrainment/impingement studies and for possible power plant mitigation (Texas Instruments, 1977a,c). The U.S. Environmental Protection Agency has indicated a desire to utilize striped bass as a test organism in national water quality standard determinations. This study was undertaken to develop laboratory culture methods.

SECTION 5

NOMENCLATURE, TAXONOMY AND MORPHOLOGY

NOMENCLATURE

Valid name

Morone saxatilis (Walbaum), Mitchill, 1814, Rep. Fishes N.Y.

Objective synonymy

Sciaena lineata Bloch, 1792, Ichthyologia, IX

Roccus striatus Mitchill, 1814, Rep. Fishes N.Y.

Roccus lineatus Gill, 1860, Proc. Acad. Nat. Sci. Phila.

TAXONOMY

Affinities

Suprageneric

- Phylum Vertebrate
  - Subphylum Craniata
    - Superclass Gnathostomata
      - Series Pisces
        - Class Osteichthyes
          - Subclass Actinopterygii
            - Superorder Acanthopterygii
              - Order Perciformes
                - Family Percichthyidae (Serranidae)

Generic

Morone Mitchill, 1814; no type description made

The generic concept adopted here is that of Whitehead and Wheeler (1966). Mitchill (1814) distinguished his genus *Morone* upon the impression that the fins were abdominal in position, in contrast to their thoractic position in the genus *Perca*. Mitchill (1814) gave no description of the genus *Morone* beyond this misapprehension of the ventral fin position.

The description is based on Jordan and Eigenmann (1890). Top of head scaly; lateral line nearly straight; teeth on tongue in one or more patches. Preopercle without antrose spines on lower limb, and lower margin simply serrate or entire. Anal spines III, 7 to 13; dorsal spines VIII-X (IX), I, 9 to 15 (12).

According to Whitehead and Wheeler (1966), Woolcott (1957), and Berg (1949), this genus contains four North American species: *Morone saxatilis* (Walbaum, 1792), *M. americana* (Gmelin, 1788), *M. chrysops* (Rafinesque, 1820), and *M. mississippiensis* (Jordan and Eigenmann, 1887); and two European species: *M. labrax* (Linnaeus, 1758) and *M. punctatus* (Bloch, 1792).

Morone Mitchill, Bleeker, 1876, 263; type *Morone americana* Gill - *Morone rufa* Mitchill.
Morone Mitchill - *Roccus* Mitchill, Boulenger, 1895, 125.
Chrysoperca Fowler, 1907; type *Morone interupta* Gill.
Lepibema Rafinesque, 1820; type *Perca chrysops*
Dicentrarchus Gill, 1860; type *Perca elongata* St. Hilaire

Specific

Morone saxatilis (Walbaum, 1792) (Figure 1)

Type locality: New York

Diagnosis: Body elongate, little to moderately compressed; back little arched; depth less than 1/3 the length, greatest depth 3.45 to 4.2, average least depth 9.6, average depth at anus 3.9--in standard length. Head subconical, 2.9 to 3.25 in standard length. Dorsal fin rays: IX (VIII-X), I, 9 to 15 (12). Anal fin rays: III, 7 to 13 (11). Ventral (pelvic) fin rays: I, 5. Pectoral fin rays: 13-19; length of pectorals 2.0 in head. The two dorsal fins are approximately equal in basal length, the first (spinous) originating over the posterior half of the pectoral, the second (soft) entirely separated from first; longest dorsal spine 2.3 in head. Anal fin situated below posterior two-thirds of second dorsal: anal spines graduated, second anal spine 5 to 6 in head. Pectorals and ventrals of moderate size; insertion of ventrals slightly behind that of pectorals. Caudal forked, the middle rays 0.6 length of outer. Lateral line scales 7 to 9-57 to 67-11 to 15; typically ctenoid. Vertebrae (including hypural): 24 or 25 (almost invariably 12 + 13 = 25). Gillrakers on first arch: 8 to 11 + 1 + 12 to 15 (10 + 1 + 14). Eye 3 to 4.9 in head and less in smaller individuals. Mouth large, oblique, maxillary extending nearly to middle of orbit, 2.5 in head; lower jaw projecting. Teeth on base of tongue in two parallel patches; also present on jaws, vomer, and palatines. Preopercle margin clearly serrate. Color olivaceous, silvery-blue; sides paler, marked with 7 or 8 continuous or interrupted blackish stripes, one of them along the lateral line; fins pale (Jordan and Evermann, 1896-1900; Merriman, 1941). (Table 1).

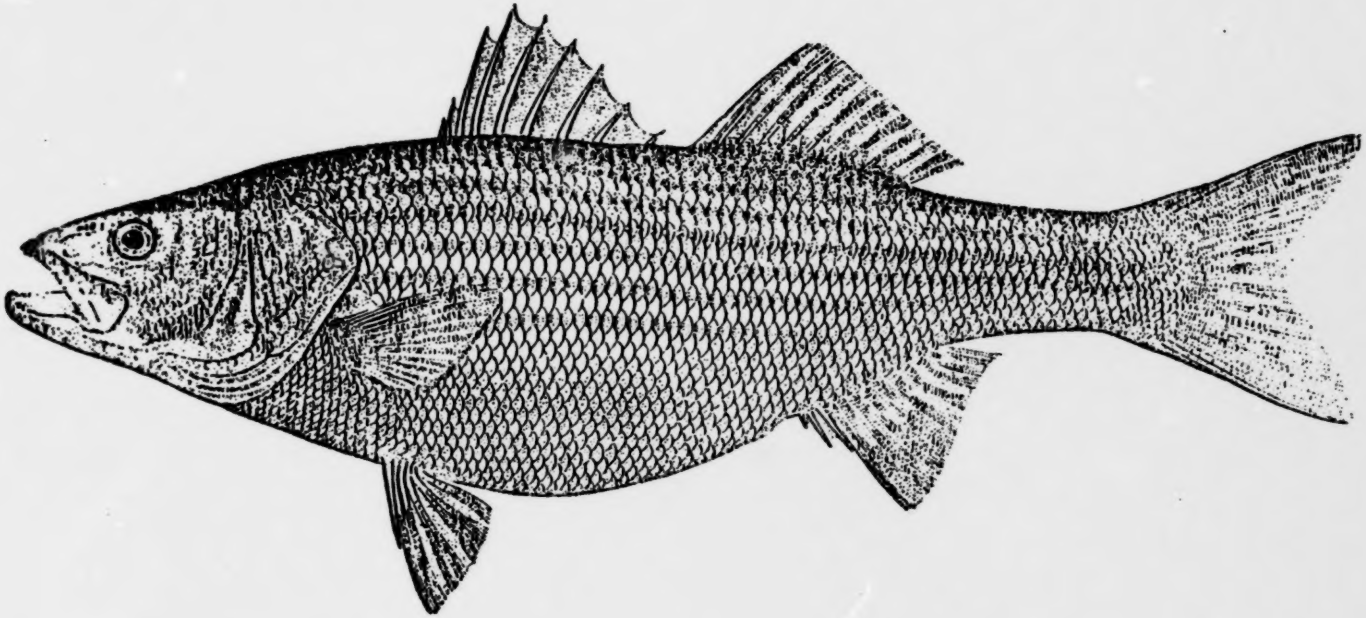


Figure 1. The striped bass, Morone saxatilis (Walbaum).

TABLE 1. DIAGNOSTIC OSTEOLOGICAL CHARACTERS OF FIVE SPECIES OF *MORONE*[*]

Character	*saxatilis*	*chrysops*	*americana*	*mississippiensis*	*labrax*
Vomer					
Dentigerous surface	Broad anteroposteriorly	Like *saxatilis*	Narrow anteroposteriorly	Like *americana*	Like *saxatilis*
Angle formed by tooth rows	Obtuse	Like *saxatilis*	Like *saxatilis*	Like *saxatilis*	Half-moon shape
Frontal	Strong. Sensory canal and pores small	Like *saxatilis*	Relatively weak. Sensory canals and pores large	Like *americana*	Like *saxatilis*
Supraoccipital crest					
Shape	Long and low	Short and high	Short and high	Like *americana*	Long and low
Length divided by height	2.0-2.5	1.5	Like *chrysops*	Like *chrysops*	2.0
Angle formed by base and dorsal margin	30 degrees	40 degrees	50 degrees	Like *americana*	35 degrees
Parietal	Not inflated	Inflated	Like *chrysops*	Like *chrysops*	Like *chrysops*
Otic region					
Area swollen	Mostly prootic	Prootic and basioccipital	Prootic, basioccipital, and exoccipital greatly inflated	Like *americana*	Intermediate between *chrysops* and *Americana*
Suture between prootic and exoccipital	Very irregular	Irregular	Entire	Like *americana*	Irregular
Otolith (sagitta)					
Shape mesially	Strongly concave	Concave	Flat	Slightly concave	Concave
Width divided by thickness at center	4.5	4.0	2.3	3.0	2.3
Length divided by width	2.5	1.7	Like *chrysops*	Like *chrysops*	2.2
Opisthotic, posterior process	Extends posteriorly	Extends dorsally	Absent	Absent	Extends posteriorly
Maxilla	Serrate at posterior edge	Like *saxatilis*	Smooth at posterior edge	Like *americana*	Like *americana*
Lower jaw	Projects beyond upper jaw. Strong. Sensory canal and pores small	Like *saxatilis*	Not projecting. Frail. Sensory canal and pores large	Like *americana*	Projects beyond upper jaw. Strong. Sensory canal and pores large
Opercle	Pronounced U-shaped notch just above hyomandibular articulation	Right angled indentation above hyomandibular articulation	Indentation absent	Indentation absent	Indentation absent
Preopercle, ventral margin	Serrate	Serrate	Serrate	Serrate	Spines (4 to 6)
Urohyal	Elongate (lateral view). Greatest depth near posterior tip. Trough lacks median ridge	Deeper than *saxatilis*. Greatest depth at distance from anterior tip. Trough with median ridge	Like *chrysops* in shape. Median ridge in trough present or absent	Like *chrysops* in shape. Trough lacks median ridge	Elongate. Greatest depth at distance from anterior tip. Trough lacks median ridge
Tongue					
Teeth on base of	Two parallel rows	Single patch	Absent	Absent	Single patch
Lateral tooth bone	Short and narrow	Long and elliptical	Long, narrow and slightly curved	Long, broad and curved	Tooth patches large and oval (bones not observed)
Dorsal pterygiophores	23	23	23	23 or 24	24
Anal pterygiophores	12	13 or 14	10	10	12
Epaxial procurrent rays	11	11	11	10	•
Hypaxial procurrent rays	11	9	9	7	•
Form of vertebral column	Straight	Slightly curved	Moderately curved	Moderately curved	Straight
10th vertebra	Haemal arch may be partially or completely formed	Like *saxatilis*	Haemal arch absent	Like *americana*	Haemal arch formed
Angle described by subcleithrum from longitudinal axis of pectoral fin	45 degrees of less	Between 45 and 60 degrees	Greater than 60 degrees	Like *americana*	80 degrees

•Not observed.

[*] Taken from Woolcott (1957), modified slightly.

Subjective Synonymy

Perca Rock-fish vel Striped Bass Schoepf, 1788, Schrift. der Gesells. nat. Freunde, VIII, 160, New York.

Perca saxatilis Walbaum, Artedi Genera Piscium, 1792, 330, New York; Bloch & Schneider, 1801, Systema Ichthyol., 89, New York.

Sciaena lineata Bloch, 1792, Ichthyologia, IX, 53, pl, 304.

Perca septentrionalis Bloch & Schneider, 1801, Systema Ichthyol., 90, pl. 20, New York.

Centropomus lineatus Lacepede, 1802, Hist. Nat. de Poissons, IV, 257.

Roccus striatus Mitchill, 1814, Rep. Fishes N.Y., 24, New York; Bean, 1884, Proc. U.S. Nat. Mus., 242, Alabama.

Perca mitchilli Mitchill, 1815, Trans. Lit. and Phil. Soc., N.Y., I, 413, pl. 3, f. 4, New York.

Perca mitchilli alternata Mitchill, 1815, Trans. Lit. and Phil. Soc. N.Y., 415, New York.

Perca mitchilli interrupta Mitchill, 1815, Trans. Lit. and Phil. Soc. N.Y., 415, New York.

Lepibema mitchilli Rafinesque, 1820, Ichthyologia Ohiensis, 23.

Labax lineatus Cuvier & Valenciennes, 1828, Hist. Nat. des Poissons, II, 79, New York; Richardson, 1836, Fauna Boreali-Americana, III, 10: Storer, 1839, Report Fishes of Mass., 7, Boston and vicinity; Ayres, 1842, Boston Jour. Nat. Hist., IV, 757, Long Island; DeKay, 1842, Zool. of N.Y. Fishes, 7, pl. 1, f. 3, Long Island; Storer, 1846, Syn. Fishes N. Am., 21; Baird, 1854, Rep. on Fishes of N.J. Coast, 7, Chesapeake Bay, Potomac, and Susquehanna Rivers; Holbrook, 1855, Ichth. S.C., 17, pl. 4, f. 2; Storer, 1855, Hist. Fishes of Mass., Mem. Am. Acad. Arts & Sci., V, 54, Mass., New Hampshire & Maine; Gunther, 1859, Cat. Fish. Br. Mus., I, 64.

Roccus lineatus Gill, 1860, Proc. Acad. Nat. Sci. Phila., 112; Gill, 1876, Ichth. Rep. Capt. Simpson's Sur. Great Basin Utah, 391; Uhler & Lugger, 1876, Md. Acad. Sci., 126; Jordan, 1878, Annals, N.Y. Acad. Sci., IV, No. 4, 97, Delaware and Potomac Rivers; Jordan & Gilbert, 1878, Proc. U.S. Nat. Mus., 380, Beaufort, N.C. and vicinity; Goode & Bean, 1879, Proc. U.S. Nat. Mus., 145, Pensacola and vicinity; Goode op cit., 115, St. John's River, Fla.; Bollman, 1886, Proc. U.S. Nat. Mus., 465, Escambia River.

Lepibema lineatum Steindachner, 1862, Verb. Zool. Bot. Ges. Wien., XII, 504.

Roccus lineatus (Bloch) Gill, Goode & Bean, 1879, Proc. U.S. Nat. Mus. 145; Jordan & Gilbert, 1883, Bull. U.S. Nat. Mus., 599.

Roccus saxatilis Jordan & Gilbert, 1883, Bull. U.S. Nat. Mus., 599; Bean, 1883, Proc. U.S. Nat. Mus., 365; Jordan, Evermann & Clark, 1930, Rep. U.S. Fish. Comm., 307.

Roccus septentrioalis, Jordan, 1885, Proc. U.S. Nat. Mus., 73.

Roccus lineatus (Bloch), Jordan & Eigenmann, 1890, Bull. U.S. Fish Comm., 423; Jordan & Evermann, 1896-1900, Bull. U.S. Nat. Mus., 1132.

Morone lineata Boulenger, 1895, Cat., I, 129.

Morone saxatilis (Walbaum), Bailey, Winn & Smith, 1954, Acad. Nat. Sci. Phila., 106, 136.

Key to the species of *Morone* (from Whitehead and Wheeler, 1966).

I. Lower border of pre-operculum with several antrorse spines; dorsal fins separated by a space; Mediterranean and Eastern Atlantic; marine and estuarine.

  a. Lateral line scales 62-74 (Mode 70); vomerine teeth in sub-crescentic band, without posterior extension; adults without black spots on upper part of body. *M. labrax* (Linn., 1758)

  b. Lateral line scales 57-65 (Mode 60); vomerine tooth patch anchor-shaped; adults with small black spots on the upper part of body. *M. punctatus* (Bloch, 1792)

II. Lower border of pre-operculum with small denticulations directed downwards; Western Atlantic, eastern & southern N. America.

  a. Dorsal fins separate; anal spines increasing evenly in length; two sharp spines on hind border of operculum; teeth on base of tongue.

    i. Body elongate, its depth more than three times in its length; lateral line scales 57-67; teeth at base of tongue in two parallel patches; marine and estuarine. *M. saxatilis* (Walb., 1792)

    ii. Body deeper, its depth less than three times in its length; lateral line scales 52-58; teeth at base of tongue in a single series; freshwater. *M. chrysops* (Raf., 1820)

  b. Dorsal fins connected; second anal spine almost equal in length to the third spine; a single sharp spine on the hind border of the operculum; teeth present along edges of tongue but not at base.

    i. Longest dorsal spine about half head length; faint streaks on flanks; marine and freshwater. *M. americana* (Gmelin,1788)

ii. Longest dorsal spine greater than half head length, seven distinct longitudinal lines on flanks, interrupted posteriorly; freshwater, lower Mississippi valley. *M. mississippiensis* (Jordan & Eigenmann, 1877)

Taxonomic status

This is a morphospecies and polytypic.

Subspecies

There are no defined subspecies.

Standard common names, vernacular names

Striped bass
Striper
Rock
Rockfish
Green-head
Squid-hound
Linesider
Missuckeke-kequoch
Rollers

MORPHOLOGY

External morphology

Some morphological data are given in the taxonomy section above.

The separation of this species into subpopulations (spawning populations) or stocks is based primarily on a morphometric study (Lund, 1957), and analysis of the variation in frequency of certain meristic counts (Table 2). Biochemical analysis (Morgan *et al*., 1973; Otto, 1975) and discriminant analysis of combinations of morphometric and biochemical characteristics have generally strengthened these separations (Grove *et al*., 1976; Berggren and Lieberman, 1978).

Lund (1957) found on the basis of morphometric analysis that a north-south cline existed along the Atlantic coast with respect to body and caudal-peduncle depth. Both measurements were lower in striped bass from Hudson-Delaware area and higher for Santee-Cooper River bass. His analysis showed that the Hudson River striped bass were distinctly different from the others he studied. Further, he concluded that, within the Chesapeake Bay, the James, York, Rappahannock and Potomac Rivers supported separate populations of bass. Lund felt that the presence of this cline suggested the differences observed were the result of selection and were, thus, genetically fixed.

Lewis (1957), counting the upper arm and total number of gill rakers on the first left brachial arch of age 0-1 striped bass, found that the Hudson River bass were homogeneous within year classes and concluded the river supported one population. He found no significant differences in gill

TABLE 2. DISTRIBUTION OF CERTAIN MERISTIC CHARACTERS AMONG STRIPED BASS SUBPOPULATIONS

Location of Subpopulation (spawning population)	Meristic Characters							
	Total Gill Rakers on First Arch (Lewis, 1957)		Lateral Line Scales (Murawski, 1958)		Dorsal Fin Rays (Raney, Woolcott & Mehring, 1954)		Anal Fin Rays (Raney, Woolcott & Mehring, 1954)	
	Range	Mean	Range	Mean	Range	Mean	Range	Mean
Rhode Island	18-25[a]	21.9	59-70[a]	63.7	10-13[a]	11.8	10-12[a]	11.9
Hudson River	22-29	25.8	53-67	60.3	9-14	11.3	9-12	10.6
Upper Chesapeake Bay	21-27	24.5	50-71	62.6	10-13	11.8	10-11	10.9
Nanticoke River	20-26[a]	22.9	59-70[a]	62.7	10-12[a]	11.6	9-12[a]	10.7
York-Rappahannock Rivers	21-26	24.0	54-67	60.7	10-13	11.8	10-12	10.9
James River	22-28	25.2	55-67	61.4	9-13[b]	11.6	8-12[b]	10.7
Albemarle Sound	22-27	24.5	56-66	60.8	11-12	11.9	10-11	10.9
Santee-Cooper System	22-26	23.5	52-64[c]	59.2	11-12	11.9	10-11	10.9
St. Johns River	---	---	52-58[c]	54.2	11-12[c]	11.9	10-12[c]	11.0
Appalachicola River	---	---	63-72[d]	66.7	10-13[d]	11.5	8-12[d]	10.5
San Joaquin River	23-29	25.9	56-64	59.5	10-11[e]	10.7	9-11[e]	10.8

[a] Authors' data

[b] Raney, 1957.

[c] Raney and Woolcott, 1955.

[d] Barkuloo, 1970.

[e] Raney and deSylvia, 1953.

rakers of 0 and I bass, but without adequate samples from bass II, he could not comment on the possibility of variation in older bass. Lewis' analysis showed that bass from western Long Island Sound had gill raker counts intermediate between those of Chesapeake and Hudson. Although Vladykov and Wallace (1952) found that vertebral counts, dorsal spines, and gill rakers were not useful characters in separating bass populations, Lewis concluded that gill rakers can be used to separate bass populations. He suggested that the possible differences found by Vladykov and Wallace in gill raker counts from five year classes within the Delaware River indicated that his samples were probably not drawn from the same population. Lewis assumed that the meristic characters he studied were genetically fixed with environmental factors operating only within narrow limits.

Murawski (1958) counted the lateral-line scales of age 0 bass from collections along the Atlantic coast. He assumed that the lateral-line scale number, once determined, does not change with age or body length. He observed that bass are about 16 mm at the time of initial formation of lateral-line scales and all have attained their full complement by the time they reach 30 mm. Murawski determined that high water temperatures during development seemed to result in low scale numbers. He felt that, to a great extent, the number of lateral-line scales in striped bass is genetically controlled, although modifiable by environmental effects, supported by his findings of a close relationship between the scale counts of Hudson and California specimens. He suggested that the within-river variations were caused by the effects of environmental changes, since they were not observed in any two succeeding year classes from a given river. Murawski concluded from his analysis of lateral-line scale variations, that the Hudson River appears to be one population (local population) of striped bass which is differentially modified by the environment. He found that the York and Rappahannock Rivers formed a homogenous group. All of the upper Bay tributaries formed another homogenous group. The James River, which lies to the south of both the York and Rappahannock Ribers, was found to have the greatest affinity with the northern Bay tributaries rather than the neighboring York and Rappahannock group. Nonetheless, he considered the James River samples as a third isolated population because it was geographically disjunct. Murawski's Delaware River samples, in turn, appeared to be most closely allied with the James River samples. In the analysis of lateral-line scale counts from Delaware River striped bass, de Sylva (1962) found no significant difference among five year classes. However, he did find a bimodality in the distribution which suggested entrance from other geographic regions.

Perhaps the most information on meristic variation in striped bass comes from counts of the soft fin rays. Raney and de Sylva (1953) reported that dorsal, anal and pectoral soft ray counts showed a significant difference between Hudson River and Chesapeake Bay bass, with the Bay usually having the higher counts of the two. They suggested that, of the three fins counted, pectorals provided best separation. However, Raney et al. (1954) found the greatest variation and difficulty in counting the pectoral fin rays. Instead they found that the soft dorsal fin rays, which appeared to be erratic in the earlier study, gave the most consistent separation, while a combination of anal and dorsal soft rays gave the highest separation. A mode of 12 soft dorsal fin-rays has been reported by Raney and Woolcott (1955), Raney et al. (1954), and Raney (1957) for striped bass from the Chesapeake region and Albemarle Sound to Mississippi, while the Hudson River appears unique with a mode of 11. Raney (1957) found the mode for soft anal fin-rays was 11 within Chesapeake Bay with 10 the next most frequent count. On the basis of soft anal fin-ray counts Raney (1957) separated the Chesapeake into two subpopulations - the James vs. the other rivers. Assuming that the meristic characters considered were genetically fixed and that environmental factors operated only within narrow limits, de Sylva (1961) concluded from his study that the Delaware River supports a spawning population of striped bass. However, since the meristic character variation predicated that the five year classes sampled were probably not drawn from the same population, he suggested this population is supplemented during some years by spawning stock from other regions, most probably from the James River.

Support of the proposed stock separations suggested on the basis of meristic characters discussed above has come from biochemical analysis. Within the Chesapeake Bay the James River appears to contain the most easily separated local population of striped bass, while the rivers north of the Rappahannock on both sides of the Bay appear to support less discrete bass populations. An electrophoretic analysis (Morgan et al., 1973) of serum proteins from adult and juveniles from five of these Upper Bay rivers, the Potomac, Patuxent, Choptank, Elk, and Nanticoke, showed four distinct populations. The Potomac and Patuxent were not distinguishable on the basis of the five proteins studied, and there appeared to be some connection between the striped bass from these rivers and the Choptank and Nanticoke bass. The Elk, the most northerly river of this study, was also the most discrete.

Otto (1975) analyzed striped bass juveniles and adults from the Maine coast, the Hudson, James, York, Rappahannock and Potomac Rivers and California (San Joaquin) to determine if there were any electrophoretically detectable differences in certain enzymes. He found that most (89.3%) of the 29 loci were monomorphic but that three (alpha-glycerophosphate dehydrogenase, isocitrate dehydrogenase, and esterase) were polymorphoric. The fraction of polymorphoric loci per population was low, ranging from zero in California samples to 8% in James River specimens. Two alleles of alpha-glycerophosphate dehydrogenase were found by Otto in all samples except those from California and the Potomac River. Significant (at 5% level) differences in esterase were found between the Hudson and James and between the James and Rappahannock specimens. Otto concluded that consistent differences between the Hudson and the aggregate Chesapeake were shown from genetic frequency of the polymorphic loci. His data indicate probable differences between the individual rivers of the Chesapeake, just as Morgan et al. (1973) found for their Upper Bay samples.

Use of meristic, morphometric and biochemical characters in discriminant analysis provides the strongest estimates for separation of three spawning stocks along the Atlantic coast. Grove et al. (1976) using discriminant functions determined from collections of adults in natal rivers (homing was assumed) from Hudson, Chesapeake and Roanoke estuaries in 1974 and 1975 were able to classify approximately 76% between the Hudson and Chesapeake spawning stocks and approximately 73% between the Hudson, Chesapeake and Roanoke stocks. The three characters providing this separation were, in order of importance, ratio snout length/internostril width, first annulus to second annulus distance/focus to first annulus distance ratio, and number of lateral-line scales. Their biochemical analysis demonstrated that the isoenzyme isocitrate dehydrogenase was fixed in the Hudson River specimens, that an isocitrate dehydrogenase allele (A) was found in Roanoke bass and not Hudson or Chesapeake specimens, and that both isocitrate dehydrogenase and alpha-glycerophosphate dehydrogenase showed a north-south clinal trend. When these biochemical data were added to discriminant analysis, the overall correct classification between estuaries increased one to two percent. Both Otto (1975) and Grove et al. (1976) agree that the biochemical genetic structure of striped bass is one of the most homogeneous (heterozygoticity of 95%) among teleosts studied.

Using only morphometric and meristic characters in discriminant analysis, Berggren and Lieberman (1978) correctly classified approximately 75% of specimens from Hudson River, Chesapeake Bay system and Roanoke River samples (1975 adults from natal rivers in these estuaries). The five characters they used, in order of importance, were ratio snout length/internostril width, scale ratio of first to second annulus/focus to first annulus, character index (as defined by Raney and de Sylva, 1953), upper arm gill raker counts, and lateral-line scale counts. These functions were then used on specimens collected from a 1975 oceanic sampling program (N = 2737) from Cape Hatteras, North Carolina, to Maine) to determine the contribution from Hudson, Chesapeake and Roanoke spawning stocks to the coastal fishery. Their analysis classified 77% of the coastal specimens as Chesapeake in origin. Separation of specimens into river populations within the Chesapeake system was not successful (Grove et al., 1976; Berggren and Lieberman, 1978).

Bryant and Seibel (1971) described tubulogenesis in striped bass mesonephros from 8 weeks to 7 years of age from freshwater impoundments. They suggested that changes in aglomerular tubules at two years of age probably reflected the confinement to freshwater. Beitch (1963) studied the urinary system to discover structures of this osmoregulatory device which might enable survival in environments of differing salinities. He found a distal tubular segment, usually present in freshwater fishes, was absent, possibly suggesting marine origin. The glomerule of a freshwater bass(55.7 μ) was found to be larger than that of a sea water captured striped bass (47.7 μ).

Protein specificity

Striped bass was one of the 30 species examined by Markert and Faulhaber (1965) to determine the variability of lactate dehydrogenase (LDH) isoenzyme patterns found in fish. They found that the bass showed three major isoenzymes (bands), no minor isoenzymes and relative migration of 0.32-0.81. A total of 31 proteins was observed in electropherograms of Chesapeake Bay striped bass serum (Morgan, Koo and Krantz, 1973). Striped bass transferrins are polymorphic and albumins are monomorphic (Morgan, 1971). However, Sidell et al. (1980) found that the transferrins are monomorphic. Three serum transferrin phenotypes of Hudson River bass are described by Hiltron (1974).

Grove et al. (1976) analyzed serum, liver, and muscle tissue using standard starch gel electrophoresis for all protein characters useful in discriminating spawning stocks. They examined 44 protein systems, including 16 serum proteins and hemoglobins. The additional 28 enzyme systems involved 52 loci of which only two were polymorphic: alpha-glycerophosphate dehydrogenase (AGPDH-1), and isocitrate dehydrogenase (IDH or ICDH-1). In addition to these two, Otto (1975) found that esterase (EST-4) was polymorphic (28 loci analyzed). Otto found variation for two loci of phosphoglucomutase (PGM-1 and 3), but the gene frequencies for the variant alleles were less than 1% in each.

Morgan (1975) distinguished larval striped bass (2 major bands) from larval white perch (3 major bands) using column acrylamide electrophoresis of the soluble muscle proteins. Sidell et al. (1978) distinguished larvae of these two species using starch gel electrophoresis and stains for specific enzyme systems. They found that the esterase (either α-napthyl-acetate or α-napthyl-butyrate) and phosphoglucomutase enzyme systems showed clear and consistent differences between the two species.

Ageing

The counting of annuli on the scales of striped bass is almost exclusively the method used for age determinations. The area of most symmetrical scales giving values for calculated length most nearly approaching the average of values from extreme body areas is that between the first and second dorsals on the second and third row above the lateral line (Tiller, 1942; Merriman, 1941). Orsi (1970) compared scales, otoliths and operculae in ageing striped bass and proposed continued use of scales primarily due to ease of handling in field collection.

Annulus formation occurs from April - June throughout the range. It occurs earlier in the southern areas and later in northern areas of the range.

Osteology

Excellent figures of the skull have been provided by Gregory (1918, 1933). The trunk skeleton was described in fine detail by Merriman (1940). Both of these authors provided the terminology used in later studies. Starks (1901) used the skeleton (including detailed skull) to illustrate the synonymy of the fish skeleton. Woolcott (1957) presented a detailed comparison of the osteology of members of the genus Roccus (Table 1). Degens et al. (1969) described the structure of otoliths from striped bass including the amino acid distribution. Daily growth rings have been reported in otoliths from 15 month old (Brothers et al., 1976) and 6 day old (Radtke, 1978) striped bass.

Frehofer (1960) illustrated the structure of Ramus lateralis system in striped bass beginning at the orbital cavity through branching of accessories I to II and inervation of pectoral and pelvic fins. His illustration is provided against background osteology.

Blood

Hematological values reported for juvenile and adult striped bass include hemoglobin values of 4.0-12.3 g/100 ml (Engel and Davis, 1964; Courtois, 1974; Westin, 1978); hematocrit values of 16-70% packed cells (Engel and Davis, 1964; Courtois, 1974; Westin, 1978); erythrocyte counts of 3.12-5.63 x 10^6/cc (Engel and Davis, 1964) and 2.86 to 4.49 x 10^6/cc (Westin, 1978). Belinsky* has recorded erythrocyte counts of 0.83-3.96 x 10^6/cc and leucocyte counts of 7-10 x 10^3/cc from yearling bass. He determined that lymphocytes accounted for 38-88% and neutrophils for 12-62% of the differential leucocyte count.

Plasma protein levels of 4.2-7.4 g% (Courtois, 1974) and 3.8-13.0 g/100 ml (Courtois, 1974; Westin, 1978) have been reported using both refractive indices and chemical methods of determination and total serum protein values using chemical methods of 3.67-8.32 g% (Westin, 1978). Serum calcium levels were reported as 9.5-15+ mg% (Courtois, 1974) and 4.5-18.8 mg% (Westin, 1978). Courtois (1974) reported values of 120-184 mEq/l serum sodium and 0.4-5.10 mEq/l serum potassium for juvenile and adult bass. Serum chloride values of 80-186 mEq/l were reported by Westin (1978) for adults. Hunn and Robinson (1966) determined plasma cholesterol to be 750 mg/100 ml (n=1), while gall bladder bile cholesterol values ranged from 1,190- 1,450 mg/100 ml (n=4).

Chadwick (1968) investigated blood lactic acid levels in netted adult striped bass. He found mean values of 630 mg/l and 1170 mg/l for those in good and poor condition, respectively.

Janssen and Meyers (1967) described an antigen against beef heart muscle present in striped bass serum but absent in white perch serum.

Hybridization

Artifically fertilized hybrids of striped bass have been successful since 1965 with crosses of striped bass females and white bass (*M. chrysops*) males at Moncks Corner, South Carolina (Stevens, 1966), and striped bass females and white perch (*M. americana*) males in North Carolina (Smith *et al.*, 1967). Poorer survival generally results when striped bass males, *rather* than females, are used in the crosses. Bayless (1968) found backcrosses of artifically fertilized hybrid striped bass x white bass males and striped bass females successful, but poor survival of white bass females and striped bass x white bass males. Bishop (1968) reported hybridization of striped bass and yellow bass (*M. mississippiensis*). Available characteristics of the striped bass hybrids are given in Table 3.

---

*Peter Belinsky, Animal Pathology, Marine Pathology Laboratory, University of Rhode Island, Kingston, RI. 02881.

No evidence has been found of natural spawning of these hybrids (Bishop, 1968; Kerby, 1972; Williams, 1972), although some mature striped bass x white bass hybrids have been observed (Bishop, 1968; Williams, 1972).

TABLE 3. MERISTIC CHARACTERISTICS OF STRIPED BASS AND STRIPED BASS HYBRIDS*

Characteristics	Striped bass	Striped bass X White perch	Striped bass X White bass
Dorsal fins	separate	connected	separate
D_2 spines	1	0-2	0-2
D_2 fin ray mode	11-12	12	13
D_2 fin ray mean		11.97	12.58
Anal fin			
spines	3	2-4	2-4
mode of rays (mean)	11	10-11 (10.54)	11 (10.96)
Lateral-line scales			
range	57-61	47-54	56-63
mean	59.70	50.00	58.67
Caudal fin rays	17	17	17
Pelvic fin spines & rays	I, 5	I, 5	I, 5
Pectoral fin rays mode (mean)		17 (16.85)	16 (16.10)
Tooth patches on tongue	2	0-2	1-2
Ratio body length to body depth	3.2:1	2.4:1	2.7:1

*taken from Bishop (1968) and Kerby (1972)

SECTION 6

DISTRIBUTION AND MIGRATION

DISTRIBUTION

The striped bass is an anadromous species occurring naturally along the Atlantic and Gulf coasts of North America (Fig. 2). It ranges from the St. Lawrence River and southern Gulf of St. Lawrence (Leim and Scott, 1968) to the St. Johns River, Florida (Merriman, 1941), and from the Appalachicola River, Florida, to the Alabama River, Alabama (Brown, 1965). In 1870 and 1882 striped bass were introduced into San Francisco Bay on the Pacific coast of North America (Mason, 1882). Their range is now from Los Angeles, California, to Barkley Sound, British Columbia (Scofield, 1931; Forrester *et al.*, 1972).

Striped bass have been established in inland freshwaters by introduction or by damming rivers (Fig. 2). Within the United States, areas with reproducing populations are Millerton Lake, California; Kentucky Lake, Kentucky-Tennessee; Kerr Reservoir, North Carolina; the Santee-Cooper Reservoir, South Carolina; and lower Colorado River, Arizona-California-Nevada. Areas of introduction with no evidence to date of reproducing stocks include freshwater ponds, lakes, rivers or reservoirs in Alabama, Arizona, Arkansas, Colorado, Florida, Georgia, Indiana, Kansas, Kentucky, Louisiana, Mississippi, Missouri, New Mexico, North Carolina, Oklahoma, Pennsylvania, South Carolina, Tennessee, Texas, and West Virginia (Bailey, 1975). Outside the United States, striped bass have been shipped to Portugal, the USSR (Stevens, 1966), and France (Delor, 1973).

MIGRATION

Local Movements

Local movements of larvae, juveniles and yearlings have been well documented in areas of proposed power plants (Hudson River, Chesapeake-Delaware Canal and Potomac River) or pump storage and canal diversions (Sacramento-San Joaquin River valley). In general, examples from Hudson River studies will be used to summarize these movements (McFadden, 1977a). Yolk-sac larvae are essentially planktonic, but appear to concentrate near the bottom at night and disperse somewhat during the day. Post-yolk sac larvae are capable of resisting currents and making more directed movements. Larvae appear to congregate near the bottom regardless of time of day and current conditions. This orientation appears to intensify as larvae approach juvenile stages. Juvenile bass are first collected in

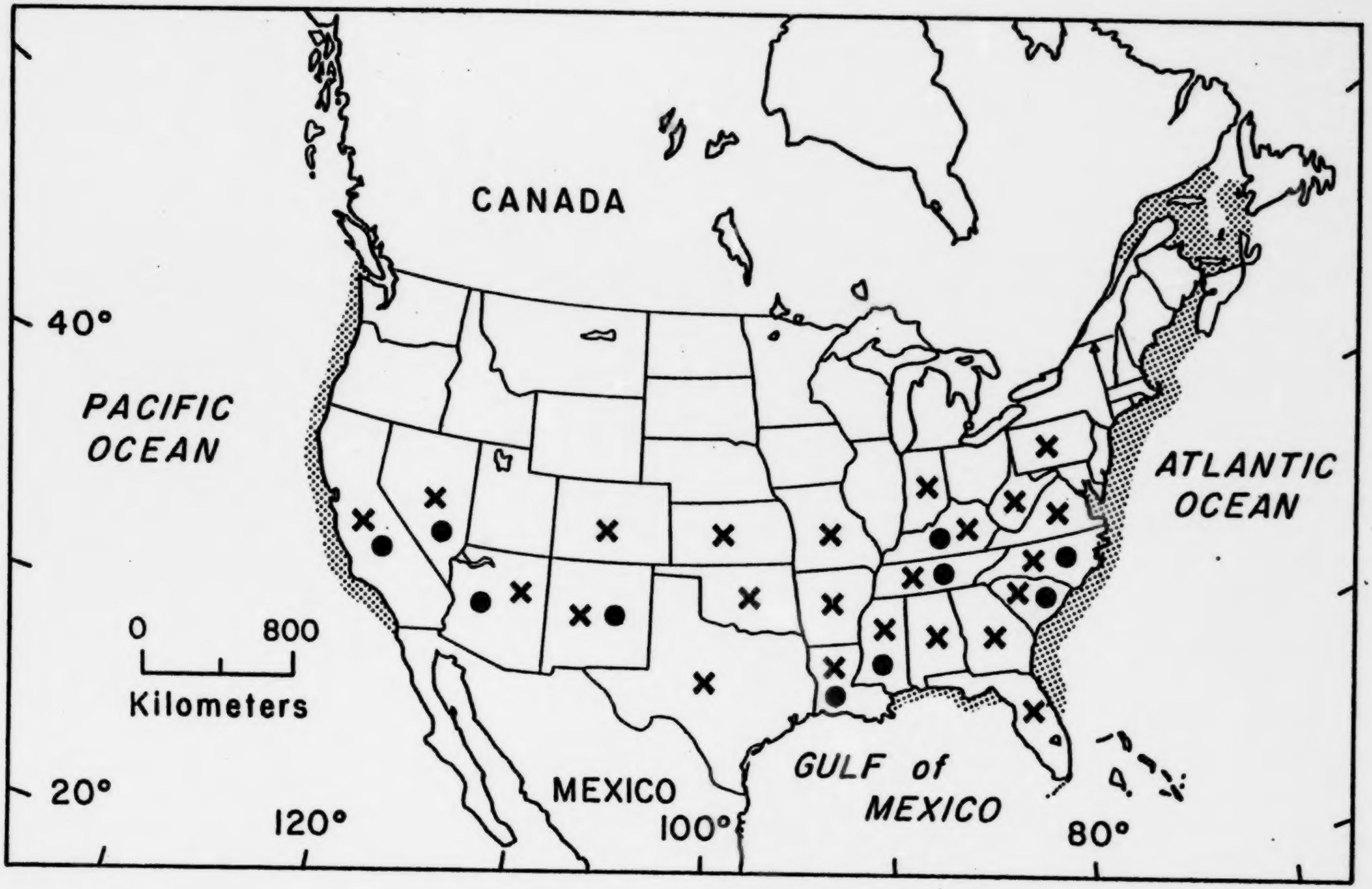

CANADA
PACIFIC OCEAN
ATLANTIC OCEAN
GULF of MEXICO
MEXICO
40°
20°
120°
100°
80°
0
800
Kilometers

Figure 2. Distribution of striped bass along the coast (stippled area) of North America and within freshwater areas of the United States (✖=stocked; ●=reproducing).

mid-June to early July, depending on time of spawning, from waters deeper than 6 meters. As water temperature increases, the juveniles migrate to shoal and shore zone areas. Falling water temperatures bring net downstream movement so that by December juveniles are generally absent from the shore zone, having either left the estuary or moved into deeper water for winter. Apparently, the abundance of juveniles in local areas is related to temperature, salinity, habitat type, diel patterns, and tidal stage. Comparisons of day/night beach seine catches in the Hudson River suggested movement into shore zone at night, probably to feed or escape predation. Yearlings are found in deep water areas in early spring, throughout the estuary by summer. With falling water temperature, they move into deeper water and downstream. Yearlings generally exhibit the same day/night pattern as juveniles, but appear less influenced by tidal fluctuations.

The migratory behavior of bass age II and over has generally been described from tagging study results. Some of these are summarized in Table 4 and are described in more detail below with other studies showing coastal migration patterns. Some segregation by size or age is evident from tagging studies. For example, investigators studying the Chesapeake Bay system seem to agree that most bass less than 30 cm (or about age III) remain within the Bay, while those over this size (age) migrate out into the coastal waters. There are also seasonal changes in migration patterns from local to coastal areas. Utilizing tag return data of the American Littoral Society and the Schaefer Saltwater Fishing Contest for bass 15 pounds (i.e., 8.6 kg; or about 80 cm FL, Table 29; or age VII-IX, Table 30) and over, Freeman (1977) concluded that there were three general groupings within the east coast distribution of this species. These are a southern (south Cape Hatteras) and a northern (north of Nova Scotia) fluviatile, and the Middle Atlantic migratory (north of Cape Hatteras and south of Nova Scotia) group.

Within the southern fluviatile group, tagging studies indicate that the migration patterns favor movement within localized areas. Scruggs and Fuller (1955) reported tagging 545 adult striped bass in the Tailrace Canal of the Santee-Cooper Reservoirs, South Carolina. Nine were recaptured during the ensuing four-month period, seven came from downstream locations in the Cooper River and two from the lower reservoir. They postulated on the basis of the latter two recaptures that there was occasionally some recruitment of bass from the Cooper River to the reservoir population.

Tagging of striped bass during 1968-70 in the Ogeechee and Savannah Rivers, Georgia, was reported by Smith (1970). In the Ogeechee River, 426 bass were tagged during the period and 103 were recovered. Of the recoveries, 25 were from upstream points and 78 were from estuarine areas of the river. A total of 259 bass were tagged in the Savannah River. Of the 43 returns, 17 came from upstream and 26 from estuarine areas. There appeared to be a general upstream movement of striped bass during the spring in both rivers just prior to spawning season. One bass tagged in the Savannah River system was recovered in the Ogeecheee. But (as Smith states) it is not known whether this bass traveled laterally between the two river systems or returned to sea to enter the Ogeechee River system

TABLE 4. SUMMARY OF TAGGING STUDIES INVOLVING AGE 2+ STRIPED BASS IN THE HUDSON, CHESAPEAKE, AND SAN JOAQUIN ESTUARIES

Area of Tagging (or location of information)	Number Tagged	Percent Return	Recapture Remarks	Source
Great South Bay, L.I., N.Y.	1,917	14.7	63% Great South Bay, Hudson River & eastern L.I.S.; 26% N.J. to Va.; 11% Conn. to Maine.	Alperin (1966a)
Great South Beach, L.I., N.Y.	580	11.6	52% South Shore L.I.; 16% R.I. & Mass.; 16% N.J. to Va.	Schaefer (1968b)
Westhampton, L.I., N.Y.	178	28.1	54% Hudson & L.I.S.; 34% N.J. to Va.; 10% R.I. & Mass.	Schaefer (1968b)
Hudson River	149	11.4	71% Hudson River & western L.I.S.; 18% South Shore L.I.; 11% Mass.	Texas Instruments (1974a)
Potomac River & north, & James River	2,869	42.1	97.5% within Cheasapake Bay; 2.5% Del. to Mass.; James River least migratory	Vladykov and Wallace (1952)
Potomac River & north	1,103	38.0	1% outside Chesapeake Bay; seasonal movements.	Mansueti (1961)
Potomac River	8,973	37.3	98% within Md. Chesapeake Bay; 1.5% Del. to Nova Scotia; seasonal movements; overwintering area for York, Rappahannock and eastern shore rivers.	Nichols and Miller (1967)
Upper Chesapeake Bay	1,762	40.9	most within Upper Bay	Moore and Burton (1975)
York, Rappahannock & James Rivers	2,429	27.8	94% within river tagged; 10 York tagged to Del. to Maine; 1 each tagged York & James to N.C.	Massman and Pacheco (1961)
Sacramento-San Joaquin Rivers	2,800	-	migrate into upper Suisan Bay to spawn and overwinter; moved into San Francisco Bay summer feeding.	Calhoun (1952)
	18,300	-	3-4 yr. olds remained in San Francisco Bay; general pattern as Calhoun (1952)	Chadwick (1967)
	7,400	-	larger bass went to sea during summer and fall; immature in freshwaters.	Orsi (1971)

later. Dudley et al. (1977) tracked 33 adult bass in the Savannah River during 1973, 1974 and 1975. They observed post-spawning movement upstream as far as 301 km from spawning areas (about 30 km upstream from the river's mouth), where the fish remained at least four months.

The migration behavior of the northern fluviatile group does not appear to be as uniformly localized as that of the southern group. Vladykov (1957) reported results of tagging in Quebec waters from 1945-1956. He observed that maximum travel of striped bass within the St. Lawrence River did not exceed 290 km. Williamson (1974) tagged 27 bass in the St. John River of which six were recaptured in Belleisle Bay, north of the tagging site, and one in Rhode Island waters. This latter bass traveled about 12 km/day before being captured. He concluded that the southern contribution to the Bay of Fundy stocks (northern fluviatile) was probably small except in years of strong year-classes. Dadswell (1976) reviewed additional tagging studies of St. John River bass. From 1964 through 1975, 280 bass were tagged (including Williamson's 27) in the St. John River. Many of these were recovered within the river, but some were recovered from Massachusetts, New York, New Jersey, Delaware and Maryland waters. In general (Dadswell, 1976), the bass tagged within the upper reaches of the St. John River remained within the estuary, while those tagged in the lower estuary were recovered there or south along the Atlantic coast. Thus, it appears that the northern stocks mix more frequently with the stocks within the Middle Atlantic, or coastal, migratory group than do the southern stocks.

Coastal Movements

Merriman (1941) provided the first major study of the movement of striped bass along the Atlantic coast. He tagged and released 2,573 bass from April 1936 to November 1937 in Connecticut and Long Island waters. By July 1938, 21% were recovered from local waters as well as waters from Massachusetts to North Carolina. Merriman concluded from his studies that there was a northern coastal migration in the spring, relatively stable localized movement during the summer, and a southward coastal migration in the fall and early winter.

Stolte (1974) reported on tag returns from a 1963 and a 1966 study of 461 bass released in Great Bay Estuary, New Hampshire. During 1963-1971, 66 of the tagged bass were recovered. One bass was recovered along the southern Maine coast and four from within Chesapeake Bay. Others were recovered within this area primarily from coastal waters, although several were recovered within Long Island Sound and the Hudson River.

Raney et al. (1954) analyzed recapture (8.5%) data from a Schaefer-Saltwater Sportsman supported tagging program from 1948 to 1952 in which most of the 9,320 bass tagged were 45 cm or less in length. The bass tagged in the Hudson River were found in the southern portion of the estuary and in the western Long Island Sound area during the summer, while they apparently spent the winter and spring in the river. Alperin (1966a) summarized a tagging study conducted from 1956 to 1961 in Great South Bay along the south shore of Suffolk County, New York. The 1,917 bass tagged were predominately ages two through four. Of the 281 recovered (14.7%), 63% were from Great South Bay and the Hudson River (eastern Long Island Sound, New York Harbor and north), 11% were from Connecticut, Rhode Island, Massachusetts and Maine waters, and 26% were from New Jersey to Virginia waters. Of the tags recovered from New York waters more came from eastern rather than western Long Island waters.

Schaefer (1968b) reported on tagging studies made from Westhampton Beach, Long Island, of 178 striped bass, from 1954 to 1956, and from Great South Bay of 4,924 striped bass, from 1961 to 1964. The bass tagged in the earlier study ranged from 30 to 60 cm FL, while over half from the latter set were over 60 cm FL. The recovery rate for those tagged from Westhampton Beach was 28.1%. Only 10% (5 bass) were recaptured north of New York (off Rhode Island and Massachusetts) and 34% (17 bass) were taken south of New York in New Jersey, Delaware, Maryland and Virginia waters. Of the 54% (27 bass) recovered from New York waters, more than half were taken from the Hudson River. During the 1961 to 1964 study 9.9% of the bass under 60 cm were recaptured, more than 75% of these were from southern Long Island waters, although one recovery was made from Maine and one from Virginia. Of the 580 bass over 60 cm tagged at Great South Beach, 67 were recaptured (11.6%) and again most (52.2%) of these were from south shore Long Island waters. Most of the others were taken in the north from Rhode Island and Massachusetts waters and in the south from New Jersey waters (about 16% each region).

Clark (1968) analyzed 1959-1963 tagging and recapture data collected by the League of Saltwater Sportsmen and found evidence of seasonal movement patterns. The Hudson River was shown to be a major spawning area and source of recruitment for bass of Long Island Sound and the New York Bight. Only three of the 195 spring recaptures were taken in the Chesapeake Bay. Hence, he concluded that bass from Long Island Sound or the New York Bight did not appear to migrate to the Chesapeake Bay to spawn. However, 72 of the 75 winter recaptures were taken from south Jersey, Delaware Bay and Chesapeake Bay, indicating probable over-wintering areas. Most of the summer and fall recaptures were made from areas off the coasts of Massachusetts and Rhode Island to New Jersey.

In a paper presented by de Sylva (Raney and Weller, 1972), the data on 309 bass collected during a 1967 to 1971 tagging program of the American Littoral Society were presented. These striped bass were either tagged or recaptured in New York waters. Those tagged along the north shore of Suffolk County and along the south shore of Long Island from Staten Island eastward appeared to be part of the coastal migratory stock since some were recovered as far south as North Carolina, Virginia, Maryland, and Delaware and as far north as Massachusetts. Six bass tagged in Maine were also from this migratory stock and were recaptured along the eastern and south shore of Long Island, and in Jamaica Bay. Raney and Weller suggest that the Atlantic coast migratory stock originates in areas as far south as North Carolina, moving north in the spring and south in the fall. This migratory stock generally moves as far north as Maine and is the basis of the Atlantic coast fishery for striped bass.

Austin and Custer (1977) used American Littoral Society tagging data for 1966-1972 to determine movements in Long Island Sound. From a total of 581 tag returns analysed, 231 bass had been tagged in the Sound but recovered outside it. Of the 350 Sound recoveries, 87% had been tagged within and 13% outside the Sound. They found most migration into and out of the Sound occurred primarily at the eastern end during spring and fall, respectively. They appear to have demonstrated an intra-sound fall migration pattern from the Connecticut to the Long Island shore before leaving the area. Bass recovered outside the Sound were recaptured to the north in waters of Massachusetts and to the south in North Carolina waters.

Tag returns of the American Littoral Society for 1971-1973 (reported in the Underwater Naturalist, 7(4) to 8(3)) were analyzed by the authors. Complete information was available for 874 tagged and recaptured striped bass (23 to 91.5 cm) during this period. Tagging was concentrated from Maine to New Jersey-Delaware, with recaptures from Maine to North Carolina. One bass, only, was reported tagged and recaptured in each of three areas - Canada, Thames River, Connecticut, and Georgia. Of the total tagged, 44% were in the Long Island Sound area, 13% were in the Staten Island area, 12% in Maine, 8.7% in Massachusetts waters, 7% along the south shore of Long Island and also New Jersey-Delaware waters, 5.5% in Rhode Island waters, and 1.5% in the Hudson River. Locations of the recaptures showed a general southerly shift. That is, 20% were recovered in Maryland-Virginia waters, 23% along the south shore of Long Island, 21% in Long Island Sound, 14% in New Jersey-Delaware waters, 6.3% in Maine waters, 4% in both Massachusetts and Rhode Island waters, 2.5% off Staten Island, 1.5% in the Hudson River, and 1% in North Carolina waters. The bass recovered in North Carolina waters came from Maine (1), Massachusetts (1), Rhode Island (1), Long Island Sound (6), and south shore (1) Long Island waters. Bass recaptured in the Hudson were tagged in Staten Island waters (6), Long Island Sound (3), and in Hudson River (4) waters. Of the bass tagged in the Hudson (13), four were recaptured in the Hudson, three at the mouth of the Hudson River, one each in Jamaica Bay and Great South Bay, three in Long Island Sound waters, and one in the Chesapeake Bay. This Hudson to Chesapeake Bay migrator was tagged in August and recovered in July of the following year. The recoveries in Maine came from Long Island Sound (2), south shore of Long Island (1), Staten Island (1), and from Maine (51) tagged bass. Of those tagged in the Staten Island area, 37% were recovered in Maryland-Virginia waters, 33% in New Jersey-Delaware waters, 17% in the Staten Island area, 5% up the Hudson River, and 0.9% (or one bass each) in Maine and Rhode Island waters.

Texas Instruments (1974a) reported on tagging returns of 592 bass 100 to 400+ mm TL released in the Hudson River during the winter and spring of 1972-1973. Only 17 of the 149 over 400 mm were recovered. The majority were recaptured within the Hudson River or western Long Island Sound, and five were recovered outside this area (two off Massachusetts and three off the south shore of Long Island).

Tagging of striped bass in southern New Jersey rivers was reported by Hamer (1971) for the period 1955 to 1970. The bass tagged were only those found in New Jersey waters for a specific reason, i.e., they were not transient bass. During 1955 to 1959, 111 wintering adult bass were tagged in the Mullica and Great Egg Harbor Rivers. The recoveries (15%) indicated that these bass migrated north into southern New England waters and returned. Spawning bass were tagged in the Maurice River from 1959 to 1970. There were 46.5% returns of these tagged bass, 24.9% from Delaware Bay and other of its tributaries, 12.5% from the Maurice River, and 4.5% from the Chesapeake Bay and its tributaries. Juvenile bass were tagged during this period in the Delaware River, with tag returns, although low, resembling that found from the Maurice River tagging.

Koo and Wilson (1972) reported on sonic tracking of 5 adult bass released in the Chesapeake and Delaware Canal during April and May, 1971. None of the bass tracked moved continuously and often the rest period was observed to be lengthy. One spent female tracked was noted to be more active at night than during the day. This behavior was not detected in the prespawning bass tracked.

Striped bass tagging in the Chesapeake Bay area began in 1931 when Pearson (1933) tagged 305 bass during July and August. Eighty-six were recaptured in the next twelve months and only nine of these were taken south of the Severn River, Maryland, the release point. To get a more complete picture over 1500 bass were tagged over a period from October 1936 to June 1937. Vladykov and Wallace (1938) reported the results of the 632 recaptures made in the nine months after tagging. This was about 42% return with 97.5% made within the Chesapeake Bay. Only eighteen bass (less than 2.5%) were taken outside the Bay and these were recaptured from February through October of 1937, from Delaware (1), New Jersey (3), Connecticut (3), Rhode Island (6) and Massachusetts (5). Vladykov and Wallace did not find a single recapture south of Chesapeake Bay. These authors tend to support Merriman's (1937, 1941) belief that the coastal migratory stock of striped bass is made up primarily of fish over three years old (1934 year-class of 1936-1937 tagging). But they did not believe that the striped bass of the Chesapeake Bay was the major contributor to this migratory stock. They felt the greater part of the Chesapeake Bay population did not move out into other bodies of water. Vladykov and Wallace also found, from tagging done in the James River, an indication of a distinct, evidently non-migratory, "school" of striped bass within the James River.

Tagging results reported by Mansueti (1961) indicated seasonal movements of striped bass in the Upper Chesapeake Bay similar to those outlined by Vladykov and Wallace (1938). Mansueti found that the recaptured (38%) bass tagged during 1957-58 remained in the upper part of Chesapeake Bay (Potomac and north), generally migrating into deeper waters during autumn for the winter, upstream with spring and returning to shallow bay waters during summer. He reported less than one percent of bass tagged were taken outside the Bay and only two were recaptured in the Virginia part of the Bay. These findings support the idea that two-three year old bass contribute little to the coastal migratory stock.

Nichols and Miller (1967) reported on a 1959-1961 tagging study during which 8,973 striped bass were tagged and released in the Potomac River and

3,345 of these were recaptured. Of the recaptures, 98% were taken in the Maryland part of the Chesapeake, only 0.5% (17 bass) were taken in the Virginia portion of the Bay, and 1.5% (52 bass) were taken outside the Bay in the Atlantic from Delaware to Nova Scotia. They include data on miles traveled and days at large for these bass. Nichols and Miller concluded that striped bass returned to the same spawning area in successive years, and that the Potomac River was a significant contributor to the coastal migratory stocks.

During the fall of 1972, striped bass 28-32 cm TL were tagged and released in the upper Chesapeake Bay. Of the 1,762 bass released, 721 (40.9%) were reported recovered (Moore and Burton, 1975). The majority of these were taken during the following winter months in deep water north of the release site. Bass recaptured during early spring were taken in the upper portions of most rivers within the Chesapeake Bay system. Six bass were recaptured outside the Chesapeake Bay.

A tagging program was initiated in 1957 to determine striped bass movements within the Virginia portion of Chesapeake Bay. During 1957-1958, Massmann and Pacheco (1961) reported 2,429 striped bass were tagged and released in the Rappahannock, York and James Rivers. Of these, 675 were recaptured and of this lot, 94% were taken in the same river system in which they were tagged. Twelve bass were recaptured outside the Chesapeake Bay. Of these, ten were from the York River taken from Delaware to Maine and one each from the York and James Rivers were taken in North Carolina waters. The James River striped bass, according to this study, moved the least of the bass from the three tagging rivers. Massmann and Pacheco suggest, as did Merriman (1941), that bass under 30 cm long do not contribute to the coastal migratory stock, but that bass over 30 cm move into the Chesapeake Bay and out along the Atlantic coast.

One study of possible migration of striped bass from North Carolina waters is Trent and Hassler (1968). They collected about 5,000 bass from the Roanoke River in the spring of 1963, 1964, and 1965. They did not report finding any striped bass tagged in northern waters. They concluded that the migratory population in the Roanoke River is composed of sexually mature bass. They feel that the population is relatively restricted to the Albemarle Sound region and possibly adjacent coastal waters. This conclusion is supported by Chapotan and Sykes' (1961) summary of tagging done from 1955 to 1959 by the United States Fish and Wildlife Service along the North Carolina coast, in Albemarle Sound, and in Chesapeake Bay. Of the 79 tagged on the North Carolina coast in 1956 and 1958, five were recaptured near the tag site, two were taken in Albemarle Sound, eight were recaptured in the Chesapeake Bay prior to and during spawning seasons, and four were taken on the Atlantic coast after the spawning season. Of the 97 bass tagged in Albemarle Sound and the Roanoke River from 1955 to 1958, five were recaptured near the tagging site, one in the Roanoke River; 16 in the Sound prior to and during the spawning season, and only one bass was taken after spawning season along the northern Atlantic coast. During the study period 206 striped bass were tagged in Chesapeake Bay tributaries and 27 were recaptured. Of the returns 12 were recovered within the Bay system, 14 were taken along the Atlantic coast, from New

Jersey to Massachusetts after the spawning season, and one was recaptured south of the Bay along the coast after the spawning season.

During 1968-1971 a total of 1,752 striped bass was tagged and released along the coast of North Carolina north and south of Cape Hatteras, with 197 returned by the end of 1971 (Holland and Yelverton, 1973). These returns indicated that three groups of bass over-winter off North Carolina. One group, mostly smaller bass, entered Pamlico and Albemarle Sounds in the spring and summer, the second, of mixed sizes, moved into Chesapeake Bay in the spring, and the third, of predominantely larger bass, moved northward during spring and summer into waters off New Jersey to Maine. It thus appears that striped bass within Albemarle Sound tend to remain there, migrating up the tributaries to spawn, while the bass along the North Carolina coast outside tend to participate in coastal migrations as far north as Maine.

In 1879 and 1882 a total of 435 juvenile striped bass were shipped from New Jersey waters and planted in San Francisco Bay. The first comprehensive study made of this stock of striped bass introduced to the west coast was that by Scofield (1931). At this time there were no tagging studies underway or initiated, therefore Scofield's conclusions were based on ecological studies only. He found a single population of bass, spawning in San Francisco Bay, which migrated along the entire California coast; that is, he distinguished them from the increasing numbers of striped bass found since 1918 in Coos Bay, Oregon. Scofield also reported that two bass had been observed in the Columbia River, 600 miles north of the Golden Gate Bridge. The only other report of the occurrence of striped bass north of Oregon was by Forrester et al. (1972). They reported the finding of two striped bass captured in British Columbia waters. One was caught in Barkley Sound and one off Port San Juan, Vancouver Island.

Tagging striped bass from California began in the early 1930's (Clark, 1934) and was limited essentially to fish three years old and less. These bass were not found to undertake definite and extended migrations (see Table 4). In 1952, Calhoun reported on results of a tag and recapture study carried out from 1947 to 1951 on over 2,800 striped bass, mostly 51 to 89 cm FL. Calhoun's report encompassed the yearly migration of striped bass into the upper delta and tributaries above Suisun Bay to spawn in early spring and in late fall for winter. Bass moved out into the Bay during late spring on their summer feeding migrations. Radovich (1963) concluded that the seaward migration of striped bass from San Francisco Bay waters was a function of coastal temperature, which might in turn effect food organism abundance. He found a positive relationship between coastal sea temperatures and seaward migration in the striped bass.

Chadwick (1967) described migration of striped bass in the Sacramento-San Joaquin River system, the two major tributaries to San Francisco Bay, based on tag returns from 13,300 bass from 1958 through 1964. In general, large mature bass migrated downstream farther than the smaller adults, and most three to four year olds remained in the Bay area during spawning season. The migration pattern from this period was found to be similar to that reported by Calhoun (1952), but with two main differences. First, in the late 1950's and early 1960's, the bass migrated farther downstream and stayed there longer than the early 1950's study revealed. Second, Chadwick found a shift from the San Joaquin to the Sacramento side of the delta, perhaps indicating an increase in the importance of the Sacramento River as a spawning area. Chadwick (1967) found that the correlation of ocean temperature and seaward migrations of striped bass as reported by Radovich (1963) did not adequately explain migration variations between 1958 and 1964.

During a 1965-1966 study in the Sacramento-San Joaquin River system over 7,400 bass, mostly mature, were tagged (Orsi, 1971). The migration pattern reported by Orsi was generally the same as that for 1958 to 1961, but showed no return to the pattern observed in the early 1950's. Only medium-sized and large fish went to sea, and then only during the summer and fall. The only bass common in freshwater during the summer were immature ones. The major differences Orsi found from Chadwick's study were a shift from San Francisco Bay to San Pablo Bay during the winter, a downstream movement of small to medium-sized bass into San Francisco Bay during the fall, a shorter residence time in the ocean during the summer, and a reduced oceanic range. Orsi felt that there seemed to be more influence on migration by bass length (age) than sex.

At the same time tagging began in California, a program was initiated in Oregon as reported by Morgan and Gerlach (1950). The majority of the 374 bass tagged from April to October 1950 were 51 cm. There were 49 bass recovered and, unlike the east coast and California studies, none were recaptured in the ocean. Their observations indicated that there were two migrations of bass within Coos Bay - an upstream spawning migration in the spring and a migration in the fall into the downstream sloughs.

Coastal migration appears, in general, to be undertaken by post-spawning striped bass of age III or over from the Chesapeake tributaries, Roanoke River and Albemarle Sound, supplemented in the Middle Atlantic and southern New England waters by bass from the Delaware and Hudson Rivers. The impressions stated by Merriman (1937, 1941) that the migrations of bass have a maximum size and intensity along New England and Long Island shores, and that the northerly spring movement is augmented by bass that have wintered farther north, appear to be supported by the more recent studies. Present indications are that bass from Albemarle Sound do not participate in the long coastal migrations of the bass from northern waters, although those off Cape Hatteras, North Carolina, may participate. Bass form South Carolina, Georgia, and Florida waters, as well as from the Gulf Coast appear to have foregone coastal migrations in favor of the fresh and brackish waters of their 'home' rivers. The Pacific Coast striped bass migrate extensively, but generally remain within San Francisco Bay and its tributaries. Coastal migrations of the nature seen on the Atlantic coast are not evident from tag returns along the Pacific coast. Another difference between Atlantic and Pacific striped bass is the direction of movement in the fall. Generally, this is into deeper, more saline waters on the Atlantic, but into the brackish-to-fresh waters of the San Joaquin Delta.

SECTION 7

MATERIALS AND METHODS

GENERAL FORMAT

The recommended culture procedures are presented by life history stage. Each stage's section includes its description, natural habitat, and environmental requirements (including biological optima) by way of introduction to the culture methodology recommendations. The procedures recommended are based in part upon data available from the literature and in part from work performed during this study.

The life history of the striped bass has been broken into four stages: embryo, larva, juvenile and sub-adult, and adult. A general description of each stage is:

embryo = spawning and fertilization to hatching;

larva including prolarva = hatching to yolk-absorption and feeding;

post larva = yolk absorption and feeding to metamorphosis;

juvenile and subadult = metamorphosis to maturity;

adult = maturity to death, including spawning.

For each stage information is organized and presented as follows in each section:

- Description of Stage
- Natural Habitat
- Environmental Requirements
  - Abiotic factors
  - Biotic factors
- Culture Methodology
  - Capture methods
  - Post-capture handling
  - Transportation
  - Handling procedures

Maintenance procedures

Culture vessels
Stocking density
Maintaining water quality
Diet

Normal conditions and physiological state.

No matter how one tries to separate each stage, there is some overlap since an individual does not grow in distinct stages, but makes a smooth transition from one to the next. For example, given the general description above, the natural habitat of a newly spawned and fertilized egg would be described twice - once from the viewpoint of the egg and again when discussing spawning adults. For clarity these areas of overlap are described in one section and the reader is referred to this description as necessary.

SOURCE OF MATERIAL

The data on the striped bass life history stages used to formulate the culture methods recommended in this report came from published reports and from studies we performed. The source of live striped bass of the various stages used for our studies was as follows:

Stage	Source of Stage	Where stage was studied
Eggs (during spawning season)	1974-1977 Moncks Corner, S.C. hatchery	1977 in S.C., 1974-1977 in R.I.
	1974-1977 Verplank, N.Y., hatchery	1975 in N.Y., 1976-1977 in R.I.
	1974-1976 Nanticoke River, Md.	1974-1975 in Md., 1976 in R.I.
Larvae (generally taken from hatching eggs on hand)	1974-1977 Moncks Corner, S.C., hatchery	1974-1977 in R.I.
	1974-1977 Verplank, N.Y., hatchery	1974-1977 in R.I.
	1974-1976, Nanticoke River,Md.	1974-1975 in Md., 1975-1976 in R.I.
Juveniles	1974-1977 eastern shore rivers, Md.	R.I.
	1974-1977 Hudson River, N.Y.	R.I.
	1975-1978 from larvae reared in lab	R.I.
Sub-adults	1974 trap fishery in Md.	R.I.
	1976-1978 from juveniles reared in lab	R.I.
Adults	1973-1976 trap fishery in R.I.	R.I.
	1975 gill-netted or rod caught in R.I.	R.I.
	1976-1978 from juveniles and subadults reared in lab	R.I.

The studies on eggs and larvae in the Hudson River, New York, and the Nanticoke River, Maryland were performed in a mobile field trailer-laboratory outfitted for this purpose. The studies on all life history stages in Rhode Island were performed at the laboratory of the University of Rhode Island's Marine Experiment Station on lower Pt. Judith Pond, Rhode Island. The research procedures used in all of our studies followed the general methods described below.

GENERAL RESEARCH PROCEDURES

All salinities were made up using filtered sea water from our flow through system (28-30 o/oo). The filters used were 5 and 10 micron cartridge or polypropylene bag (GAF) filters. In the laboratory the source of freshwater was dechlorinated tap water, while in the field the source of freshwater was filtered (5 µ bag) river water. Juvenile, subadult and adult bass populations were maintained in ambient flow through sea water systems in the laboratory until used for feeding and metabolism studies.

A number of egg and larval studies were performed under constant temperature regimes. Constant temperatures of 12, 15, 18, 21, 24, and in some cases 27°C were maintained in test containers by keeping them immersed in a temperature controlled water bath. Bath temperatures were controlled using Haake 1000 watt heater thermo-regulators operating against a cooling coil in each bath. Other studies were performed at ambient sea water temperatures. Figure 3 shows average year-round ambient sea water temperatures at the laboratory.

Water quality was monitored throughout these studies. Dissolved oxygen was determined using a Yellow Springs Instruments Co. (YSI) dissolved oxygen probe, supplemented periodically with determinations using the azide-modification of the Winkler titration. The pH was measured using an Orion pH electrode. Ammonia was determined using a micro-modification (1/50th reduction in sample and reagents) of the indophenol technique of Solorzano (1969). Salinity measurements were made using an American Optical salinity refractometer. Conductivity measurements were made with a Y.S.I. conductivity bridge and a one cm cell.

Caloric values of egg constituents, empty representative bass, two diets and the feces produced by bass consuming each diet were determined using a Parr adiabatic bomb calorimeter. Benzoic acid tablet standards were run concurrently. Residue, or ash values were calculated from these determinations. These were checked against ash values obtained directly from igniting subsamples of material in a muffle furnace at 450°C for five hours. Percent carbon, hydrogen and nitrogen content of larvae, two diets and their feces was calculated from analysis performed on the EPA Narragansett Laboratory's Carlo-Erda (model 1100) analyser.

Feeding larvae were supplied with newly hatched *Artemia* nauplii at least twice a day in quantities sufficient so that a portion remained at the next feeding. *Artemia* nauplii proved to be a satisfactory diet for striped bass through the early juvenile stage. Juveniles and yearlings were fed cut or ground squid or a moist diet described in the juvenile

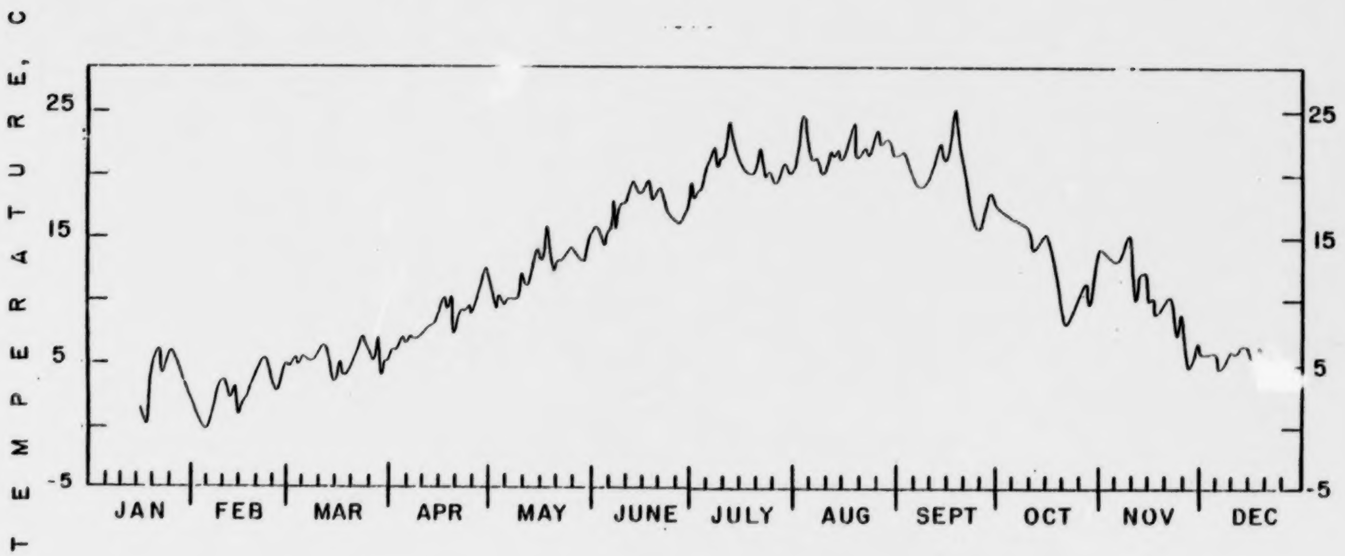


Figure 3. Yearly ambient sea water temperatures from flow through laboratory holding tanks during 1974.

section. Sub-adults and adults were fed cut squid or menhaden.

Egg and larval sampling procedures and weight length measurement determinations are described in Rogers (1978), Rogers et al. (1977), and Rogers and Westin (1979). In the embryo and larval sections there are areas where data is reported as 'typical'. This refers to one of two to four replicates (or an average) of an experiment performed at different times, often in different years.

Groups of juveniles (5-140 g wet weight) were used in experiments to determine food consumption, feeding frequency, evacuation rates, oxygen consumption, ammonia excretion, and growth. The bass used in these studies were distributed into Frigid Unit fiberglass oval tanks (150 l) according to their size (weight) so that the largest fish in a particular tank was within 2.0 times the size of the smallest fish. Each tank was supplied with aerated flowing filtered seawater (28-30 o/oo) at a replacement rate of about a liter per minute. The water temperature in each experiment was recorded daily. The bass were weighed on day one and again at the end of the experimental period. Although the fish were weighed separately, the individual weights from a particular tank were combined to calculate the group weight. When weighed, each fish was removed from the tank and anesthetized using quinaldine (0.01 ml quinaldine to 1.0 liter water). Each fish was then blotted dry and both the fork length and wet weight (to the nearest 0.1 gram) were measured. The fish were returned to sea water immediately after measuring. They were not fed on the day they were weighed.

The primary food items chosen for use in feeding studies were obtained commercially or made from commercially available materials. The fresh frozen foods (squid and menhaden) were cut into pieces that were easily eaten by the striped bass. The menhaden were headed before being used for food. The bass were fed to satiation daily and the rations weighed to the nearest gram. Evacuation rates were estimated from returns of colored hobby store beads (2 mm diameter) put into food pieces prior to feeding. The number of beads consumed (uneaten portions were removed) was recorded. As the beads were evacuated, they were collected in a sieve during the siphoning of the bottom of the tank three times a day. The length of time for the beads to evacuate was recorded in hours and the number of beads that returned in each tank was calculated as a percent of the total. The percent was plotted against time to evacuate and a graphic estimate was made. A second estimation was made by transforming evacuation rates to probits and hours to log scale. A linear regression of probits on time in hours was then calculated for each group at each temperature period in which there were four or more points from 10-90% returned.

The oxygen consumption measurements of bass over 200 g wet weight were made in 70 liter tunnel respirometers, or in clean, darkened (covered) 500 liter fiberglass tanks. Tunnel respirometers (NMFS, Milford Laboratory) were used to determine the oxygen consumption of bass 200 to 500 g wet weight at 15°C and 19°C. The measurements in the tunnel respirometers were made at velocities of 20 to 80 cm/sec using methods described by Freadman

(1978). Standard metabolism of each fish was estimated from this data by extrapolating to 0 cm/sec. Oxygen consumption of bass > 500 g made in the 500 l tanks and on those < 200 g were measurements of routine metabolism. All routine metabolism measurements were made after two days acclimation for two hour periods. Appropriate controls and initial oxygen levels were measured. The initial oxygen levels were not less than 90% of saturation. Oxygen levels were not allowed to fall below 55% of saturation during a measurement period. In both types of respiration determinations, the bass were starved for 24 to 36 hours prior to testing unless otherwise indicated.

Excretion was measured as ammonia-nitrogen in both freshwater (dechlorinated tap water) and seawater. Measurements were made concurrently with respiration for those fish greater than 1000 g live weight. Other determinations were made on individual fish independent of the respiration measurements. All fish were starved 24-26 hours during the acclimation period before the initial measurements were taken. Additional measurements were made daily thereafter. Food was withheld during these measurement periods.

The data obtained from the initial and final weighings of each bass group and the recorded amount of food consumed by the fish during the experimental period were used to calculate the growth rate and gross growth efficiency of the groups of fish using the following formulas:

$$\underset{(\%)}{\text{Gross Growth}}\ \text{Efficiency} = \frac{\text{Weight Gain}}{\text{Total Consumed}} \times 100$$

$$\begin{array}{l}\text{Growth Rate}\\ \text{(\% per day)}\end{array} = \frac{\dfrac{\text{Final Dry Weight - Initial Dry Weight,}}{\text{Initial Dry Weight}}}{\text{\# Test Days}} \times 100$$

These calculations of growth were either on a dry-dry or wet-wet weight basis. This is indicated when the values are given.

Some striped bass ovarian and muscle tissues were sampled for organochloride concentration. The frozen samples were thawed and analysed in lots of five plus reagent blanks using facilities in Dr. Charles Olney's laboratory (URI). The thawed tissue was ground with sodium sulfate and petroleum ether to extract the lipid material and the contaminants. Alumina clean-up was used to remove the lipid material. Following clean-up, the silicic acid separation methods described by Bidleman et al. (1978) was used. The samples were analysed on a gas chromatography (Tracor MT-220) with Ni^{63} electron capture detectors. The chromatograph had columns of 1.5% OV-17 / 1.95% QF-1 and 4% SE-30/6% QF-1 and was operated at 200°C. The PCB and DDT (DDD, DDE and DDT) dieldrin, and chloradane concentrations in the tissues were calculated against appropriate reference standards.

In addition to any background references given in each section, there are a number of works with which we have assumed the reader is familiar. These include the Fish Physiology series of eight volumes edited by W. S. Hoar and D. J. Randall and Fish Nutrition edited by J. E. Halver. Two very good reviews on nutrition requirements of fish have been assembled by the National Research Council (1973, 1977) for their Nutrient Requirements of Domestic Animals Series. Spotte (1979) provided an extensive background on water quality control in closed-system rearing environments. In addition, Amlacher (1970), Klontz (1973) and Kingsford (1975) were very useful in dealing with tentative disease or problem diagnosis among our laboratory populations.

SECTION 8

RECOMMENDED CULTURE METHODS AND BIONOMICS: EMBRYO

DESCRIPTION OF STAGE

This stage emcompasses that portion of the striped bass life history from spawning and fertilization to hatching.

Striped bass eggs are semibouyant with a large perivitelline space. Diameters of fully water-hardened fertilized eggs measured live range from 1.25 to 4.50 mm. Figure 4 shows the range of chorion diameters from a number of spawning areas. The areas consisting mainly of smaller diameter eggs (Blackwater and Transquaking Rivers) have predominately higher salinities (2-3 o/oo) during the spawning season (Hollis, 1967). Albrecht (1964) found egg specific gravity was related to egg size, i.e., smaller eggs have a slightly higher specific gravity. He reported the average specific gravity of striped bass eggs to be 1.0005, with a range of 1.0003 to 1.00065.

Each egg has one amber-colored oil globule, which has been noted to fragment (Mansueti, 1958), and a single yolk. Yolk and oil diameters are shown in Figure 4 for live water-hardened eggs.

The caloric content of striped bass eggs has been reported to be an average of 8,031 cal/g (Rogers, 1978) and 8,070 cal/g (Eldridge et al., 1977). The percent of whole egg dry weight that each constitute (namely chorion, yolk, and oil) comprises is summarized in Table 5 from data given in Rogers (1978). Eldridge et al. (1977) reported that their eggs averaged 2.21 calories per egg and contained oil of 9291 calories per gram. Carbon: nitrogen determinations on groups of ten whole unfertilized eggs revealed approximately 7% nitrogen and 48% carbon (Rogers, 1978). During the course of this study unfertilized eggs were obtained from 36 gravid females of known length (and usually weight) from one of three locations. Dry weight determinations were made on six to eight groups of 50 eggs from each female. This data is presented in Table 6. It suggests that larger females produce larger, or heavier, eggs.

The developmental stages of striped bass eggs are shown in Figure 5. The rate at which this development proceeds depends on temperature (Figure 6). The hatching time of eggs in relation to water temperatures is summarized in Table 7. The time to hatch was estimated by Rogers et al. (1977) as

$$\text{time to hatch (hours)} = 258.5e^{-0.0934\ (^{\circ}\mathrm{C})}$$

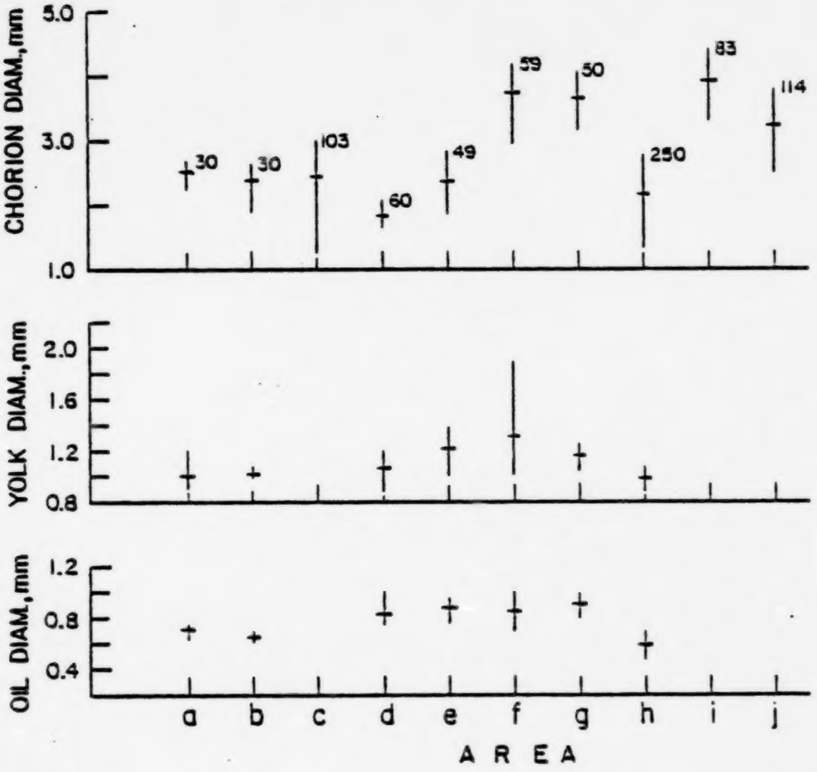


Figure 4. Regional variation in striped bass egg dimensions; chorion, yolk and oil diameters measured in live material from several locations indicated by letters. Number to right refers to the number of eggs measured from each location.

a - Hudson River, hatchery 1975 (Rogers, 1978)
b - Hudson River, hatchery 1976 (Rogers, 1978)
c - Delaware River (Bason, 1971)
d - Blackwater River, Maryland, 1974 (Rogers, 1978)
e - Transquaking River, Maryland, 1974 (Rogers, 1978)
f - Nanticoke River, Maryland, 1974 (Rogers, 1978)
g - Choptank River, Maryland, 1975 (Rogers, 1978)
h - South Carolina, Moncks Corner Hatchery, 1976 (Rogers, 1978)
i - San Juaquin River, California, May 1962 (Albrecht, 1964)
j - San Joaquin River, California, June 1962 (Albrecht, 1964)

TABLE 5. SUMMARY OF THE ENERGY CONTENT OF UNFERTILIZED STRIPED BASS EGGS

Egg Component	Mean percent of whole egg dry weight	Calories per mg dry weight	Calories per 0.300 mg dry egg
yolk (less ash)	36.48 (160)	5.75 (7)	0.63
oil (less ash)	51.68	10.89 (6)	1.69
chorion (less ash)	8.17 (160)	5.65+	0.14
ash	3.37	-	-
whole egg (calculated)	99.70		2.45
whole egg (direct calorimetry)	100.00		2.41 (7)

+ Not measured directly in this study. Caloric value for protein were used (Phillips, 1969) since the chorion is probably protein.

* Numbers in parenthesis refer to sample size. Mean percent oil of whole egg was determined by subtraction.

TABLE 6. RELATIONSHIP BETWEEN THE SIZE OF GRAVID STRIPED BASS FEMALES AND THE DRY WEIGHT OF THE EGGS THEY PRODUCE (ROGERS, 1978)

Identification	Fish Size		Mean Egg Weight per 100 Eggs (mg)	Range in Egg Weight (mg/100 eggs)	Standard Deviation
	Fork Length (cm)	Weight (kg)			
New York, 1975					
Roe 5	88.7	9.75	31.40	30.8-32.6	0.0068
Roe 6	93.2	12.02	39.27	37.8-40.2	0.0086
Roe 7	89.5	7.26	29.90	28.6-31.8	0.0135
Roe 15	54.6	6.93	31.97	30.0-31.8	0.0025
New York, 1976	115.0	15.20	25.60	25.0-26.4	0.0064
Maryland, 1975					
4/25-1a	102.9	14.50	38.00	37.0-40.0	0.0119
			35.67	33.2-36.8	0.0129
			37.62	36.8-38.8	0.0076
4/25-1b	107.9	17.01	37.20	35.6-39.2	0.0132
			36.83	36.4-37.6	0.0048
			36.97	35.6-37.8	0.0078
4/28-2	84.5	8.39	34.63	32.4-36.4	0.0150
4	118.0	23.59	37.73	36.6-40.0	0.0127
7	89.4	8.98	28.23	22.2-38.4	0.0788
Maryland, 1976					
A	99.0	15.88	46.60	45.8-47.6	0.0070
B	68.0	3.29	28.67	28.0-29.4	0.0048
D	98.0	14.06	40.03	39.4-40.6	0.0041
E	108.0	16.33	43.03	41.0-49.0	0.0257
F	108.0	11.34	37.13	36.4-37.6	0.0045
G	79.0	5.44	29.53	29.2-30.4	0.0048
			30.47	29.6-31.0	0.0058
6	71.0	4.99	28.73	28.6-29.0	0.0016
9	64.0	3.29	19.96	19.0-20.6	0.0054
South Carolina, 1976					
Roe 57	77.0		26.87	26.2-27.8	0.0059
Roe 58	67.0		22.35	21.8-23.1	0.0049
Roe 59	77.5		22.55	21.8-23.5	0.0061
Roe 60	74.0		20.00	19.2-20.6	0.0051
Roe 63	78.0		27.23	26.8-27.8	0.0039
Roe 64	74.0		20.47	19.4-21.2	0.0063
Roe 65	71.0		15.60	15.2-16.0	0.0028
Roe 66	71.0		23.03	22.6-23.4	0.0029
Roe 67	73.5		22.97	21.6-23.8	0.0082
Roe 68	72.0		22.43	21.8-23.2	0.0053
Roe 69	74.0		27.00	25.6-29.0	0.0125
Roe 70	71.0		26.60	26.2-27.0	0.0033
Roe 71	76.0		29.40	28.2-30.2	0.0075

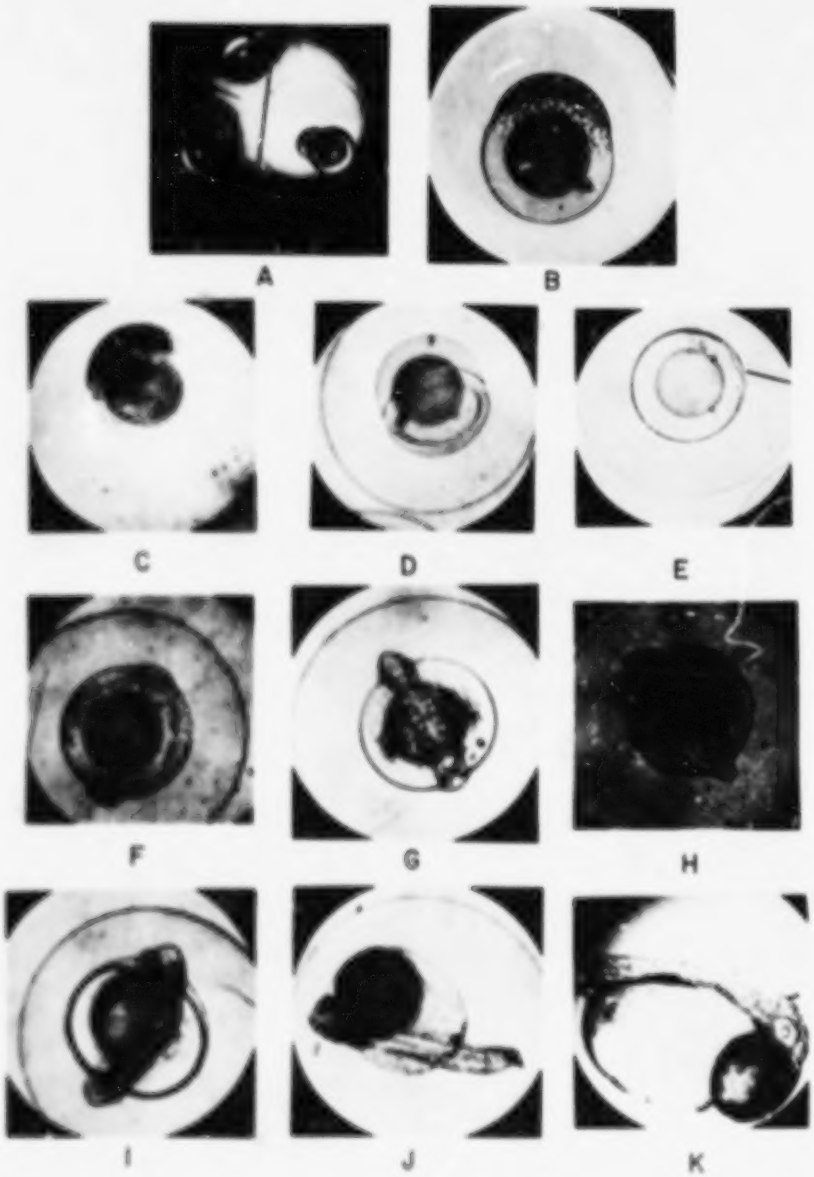


Figure 5. Development of striped bass eggs at 18.8-20°C. (after Bayless, 1972)

A - fertilizated egg at 2 hours, note cleavage (20X)
B - 5 hours (50X)
C - 10 hours (50X)
D - 12 hours (50X)
E - 18 hours, ventral view (50X)
F - 21 hours (50X)
G - 24 hours (50X)
H - 28 hours (50X)
I - 32 hours (50X)
J - 36 hours (50X)
K - 44 hours, hatching (50X)

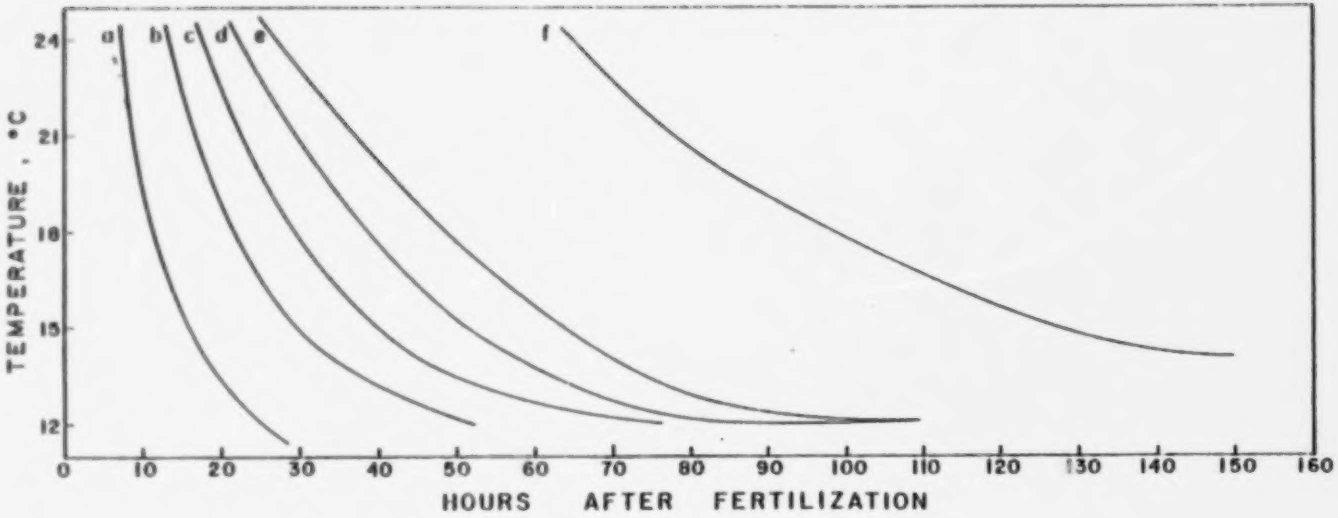


Figure 6. The effect of incubation temperature on the time from fertilization to selected developmental stages before and after hatching (Rogers et al., 1977).

a - half of yolk enveloped by the blastoderm (Figure 5D)
b - embryo extending over half of the yolk (Figure 5F)
c - early tail development (Figure 5H)
d - free tail bud (Figure 5I)
e - hatching (Figure 5K)
f - development of eye pigmentation in the prolarva

TABLE 7. HATCHING TIME OF STRIPED BASS EGGS IN RELATION TO WATER TEMPERATURES

Incubation Time (hours)	Water Temperature (°C)	Location	Author
25	26.67	N.C.	Shannon and Smith (1967)
25.8	24.00	N.Y.	Rogers *et al.* (1977)
28	23.89	N.C.	Shannon and Smith (1967)
28.5	24.00	N.Y.	Rogers *et al.* (1977)
30	23.33	N.C.	Shannon and Smith (1967)
30	23.33	S.C.	Bayless (1972)
30	21.7-22.2	N.C.	Bigelow and Schroeder (1953)
30	21.7-22.2	-	Merriman (1941)
33	21.11	S.C.	Stevens (1965)
33	21.1	N.C.	Regan *et al.* (1968)
34	21.11	N.C.	Shannon and Smith (1967)
35	22.22	S.C.	Bayless (1972)
36	21.67	N.C.	Worth (1884)
37	21.00	N.Y.	Rogers *et al.* (1977)
36-48	17.22	N.C.	Mansueti (1958)
38	19.4	N.C.	Regan *et al.* (1968)
38	21.11	S.C.	Bayless (1972)
40	20.00	S.C.	Bayless (1972)
43	18.3	N.C.	Regan *et al.* (1968)
44	18.33	S.C.	Stevens (1965)
44	18.89	S.C.	Bayless (1972)
48	19.4	N.C.	Bigelow and Schroeder (1953)
48	18.33	S.C.	Bayless (1972)
48	17.2	N.C.	Regan *et al.* (1968)
48	17.89	-	Pearson (1938)
48	18.89-19.44	N.C.	Worth (1882)
50	15.6	N.C.	Regan *et al.* (1968)
50	17.78	S.C.	Bayless (1972)
51.8	18.00	N.Y.	Rogers *et al.* (1977)
54	14.4	N.C.	Regan *et al.* (1968)
56	16.67	S.C.	Bayless (1972)
58	15.56	N.C.	Shannon and Smith (1967)
62	15.00	N.Y.	Rogers *et al.* (1977)
62	15.56	S.C.	Bayless (1972)
66.3	18.00	N.Y.	Rogers *et al.* (1977)
70	15.56	S.C.	Stevens (1965)
70-74	14.4-15.6	N.C.	Bigelow and Schroeder (1953)
74.3	15.00	N.Y.	Rogers *et al.* (1977)
74	14.4-15.6	-	Merriman (1941)
74	14.44	Md.,Va.	Brice (1898)
91.8	15.00	N.Y.	Rogers *et al.* (1977)
109	12.00	N.Y.	Rogers *et al.* (1977)

NATURAL HABITAT

The water temperature on striped bass spawning grounds during the spawning season has been reported as low as 8°C (Westin, 1978) and 10°C (Carlson and McCann, 1969) early in the season, and as high as 23°C (Carlson and McCann, 1969) and 25°C (Scruggs, 1957) near the end of the season. Table 8 summarizes data on striped bass spawning determined from egg collections. The season occurs during late March to late June depending on the time of spring warming. Major spawning peaks generally occur as the water temperatures reach 14-16°C. The salinity during the spawning season has been reported as fresh to brackish; that is, from 0 to 10 o/oo (Turner and Farley, 1971), 0 to 3 o/oo (Dovel, 1971), 0 to 5 o/oo (Tresselt, 1952) and 0 to 10 o/oo (Hollis, 1967). Tresselt (1952) determined that most spawning activity occurs within the first 25 miles of freshwater in the spawning river. Our measurements during the 1975 spawning season on the Nanticoke River, Maryland, plotted in Figure 7 show the temperature, turbidity, and salinity ranges (600 μmhos < 1 o/oo). The distribution of striped bass eggs in the Hudson River with temperature is shown in Figure 8.

Spawning grounds are generally turbid and usually in an area of good current or tidal flow. Turbidity during the 1975 spawning season on the Nanticoke River, Maryland ranged from 17 to 46 JTU and appeared inversely related to temperature. Turbidity as high as 132 JTU was reported by Smith (1970) during 1968 spawning in the Savannah River, but only 11 to 80 JTU during the 1969 spawning. McCoy (1959) reported river discharges of 5,500 to 30,225 cfs (1.5 to 3.0 fps) on the Roanoke River spawning grounds at Weldon, North Carolina. On the Tar River spawning ground, Humphries (1966) recorded river discharges of 700 to 4080 cfs. The mean inflow for the San Joaquin River, California during the 1948 spawning season ranged from 1367 to 4533 cfs (Erkkila et al., 1950). Tresselt (1952) recorded a range of 0.6 to 2.0 fps for the Virginia spawning grounds he studied.

ENVIRONMENTAL REQUIREMENTS

The environmental requirements of striped bass embryos are summarized in Table 9. These requirements have been separated into the abiotic and biotic factors and are discussed more fully below.

Abiotic Factors

Optimal temperatures for embryonic growth and survival have been reported ranging from 14 to 24°C (Albrecht, 1964; Bayless, 1972; Morgan and Rasin, 1973). However, survivals at 15 and 18°C were higher than those of eggs reared at 12 or 24°C, and 18°C was proposed as optimal for embryonic development (Rogers et al., 1977). An upper lethal temperature of 27°C and a lower lethal of 11°C have been suggested (Morgan and Rasin, 1973). Shannon (1970) found that the longer egg development was maintained at 18°C, the more tolerant the eggs became to shock exposure to higher temperatures. Koo and Johnston (1978) exposed early eggs to ΔT's of 10 and 15°C above an 18°C base temperature for exposure periods of 5 to 180 minutes. They observed reduced hatchability with an increase in numbers of deformed

TABLE 8. DATA ON STRIPED BASS SPAWNING THROUGHOUT ITS RANGE

Spawning River	Time of Spawning	Temperature (°C)	Date and Temp. (°C) at Peak of Spawning	Source
Hudson River, N.Y.	24.IV-25.VI 1966	10-22.2	29.V;16.1	Carlson and McCann (1969)
	7.V-25.VI 1967	10-22.8	21,29.V;13.3	
	21.IV-30.VI 1968	11.7-22.8	12-18.V;15.6	
Delaware River, Chesapeake-Delaware Canal, Delaware	28.IV-4.V 1970	13-14	—	Bason (1971)
Nanticoke River, Maryland	14.IV-23.V 1960	14.3-20.7	15.IV;15.0	Hollis (1967)
	28.III-27.V 1964	11.4-23.9	20.IV;16.7	
	11.IV-10.V 1975	8.0-17.2	—	Westin (1978)
Potomac River, Maryland	21.IV-26.VI 1975	10.9-23.4	28.IV;14.3	Hallowing Point Field Station (1976)
Rappahannock River, Virginia	17.V-20.V 1950	19.4-20.4	—	Tresselt (1952)
Mattaponi River, Virginia	25.IV- 1950	13.9-20.5	30.IV;16.8	Tresselt (1952)
Pamankey River, Virginia	6.IV- 1950	13.4-14.3	13.IV;13.0	Tresselt (1952)
	13.IV-10.V 1966	15-22	—	Rinaldo (1971)
Chickahominy River, Virginia	5.V-6.V 1950	18.7-20.4	—	Tresselt (1952)
James River, Virginia	—	—	9-10.V;19-21	Tresselt (1952)
Roanoke River, North Carolina	11.IV-10.V 1938	21-25	—	Merriman (1941)
	14.V-2.VI 1958	16.2-19.7	25.V;18.3	McCoy (1959)
	21.IV-9.VI 1967	16.7-18.9	—	Hassler _et al_. (1970)
	26.IV-2.VI 1968	15-21	—	"
Tar River, North Carolina	14.IV-18.V 1965	15-22	3-11.V;15-18	Humphries (1966)
	21.IV-20.V 1975	14.5-21.1	5-12.V;17.8-18.4	Kornegay&Humphries (1976)
Congree River, South Carolina	8.IV-2.VI 1955	16.1-25	21.IV	Scruggs (1957)
Wateree River, South Carolina	8.IV-19.V 1955	15-22.2	5.V	Scruggs (1957)
Diversion Canal, Lake Marion, S.C.	6.IV-11.V 1955	14.4-24.4	27.IV	Scruggs (1957)
Ogeechee River, Georgia	2.IV-23.IV 1968	19.4-21.7	—	Smith (1970)
	1.IV-12.V 1969	17.8-23.3	—	Smith (1970)
Savannah River, Georgia	31.III-29.V 1969	16.7-22.2	—	Smith (1970)
Sacramento River, California	11.IV-29.VI 1964	16.1-20.6	29.V;20.6	Farley (1966)
	2.V-20.VI 1973	15.6-20	10.V;18.3	California (1974)
San Joaquin River, California	5.IV-17.VI 1949	16.6-22.2	—	Erkkila _et al_. (1950)
	1.IV-10.VI 1957	15.6-21.1	29.V;18.3	Chadwick (1958)
	11.IV-29.VI 1964	13.3-21.1	—	Farley (1966)

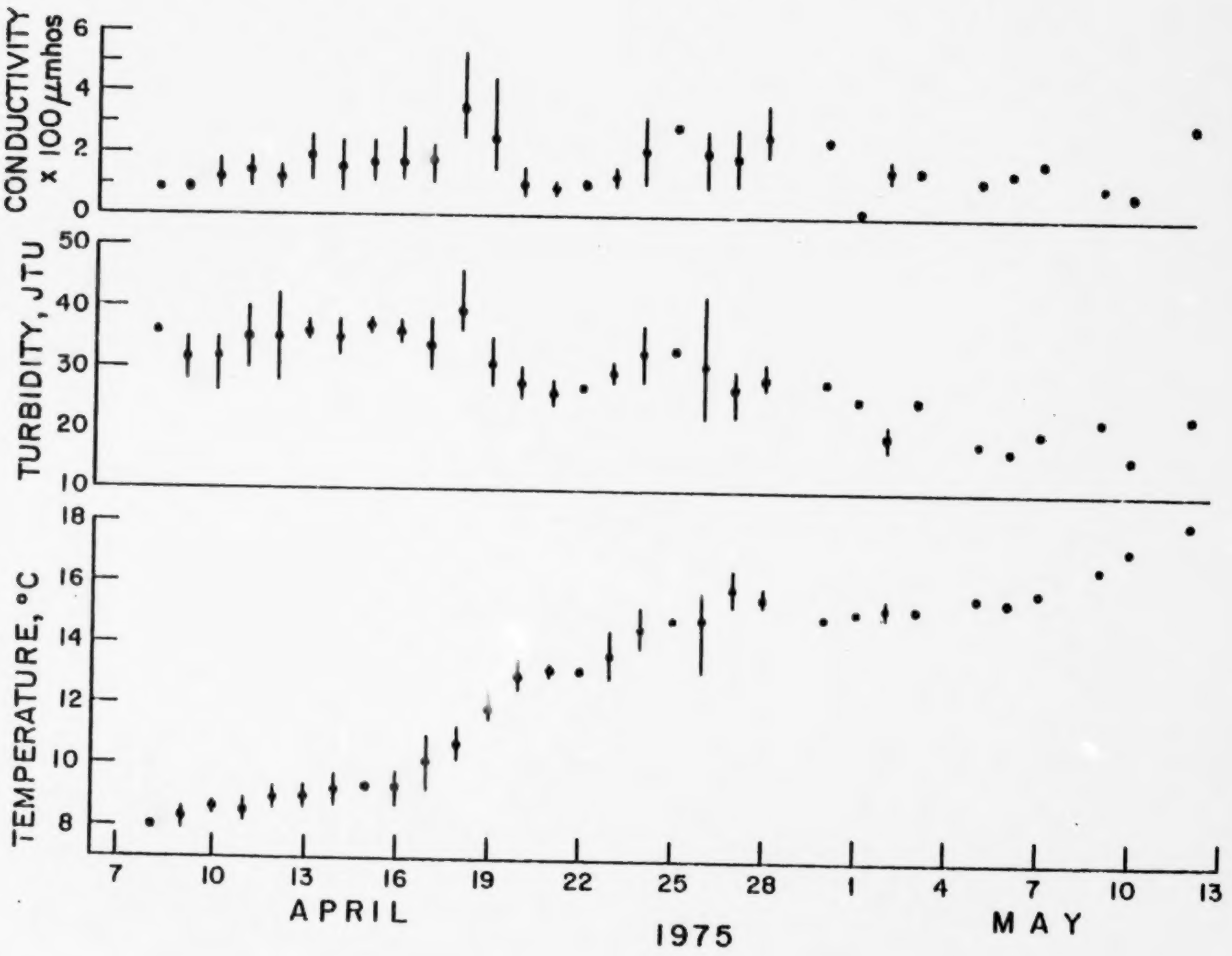


Figure 7. Temperature, turbidity, and conductivity measured on the spawning grounds of the Nanticoke River, Maryland, during 1975. Vertical bar is the range for a given day's measurements.

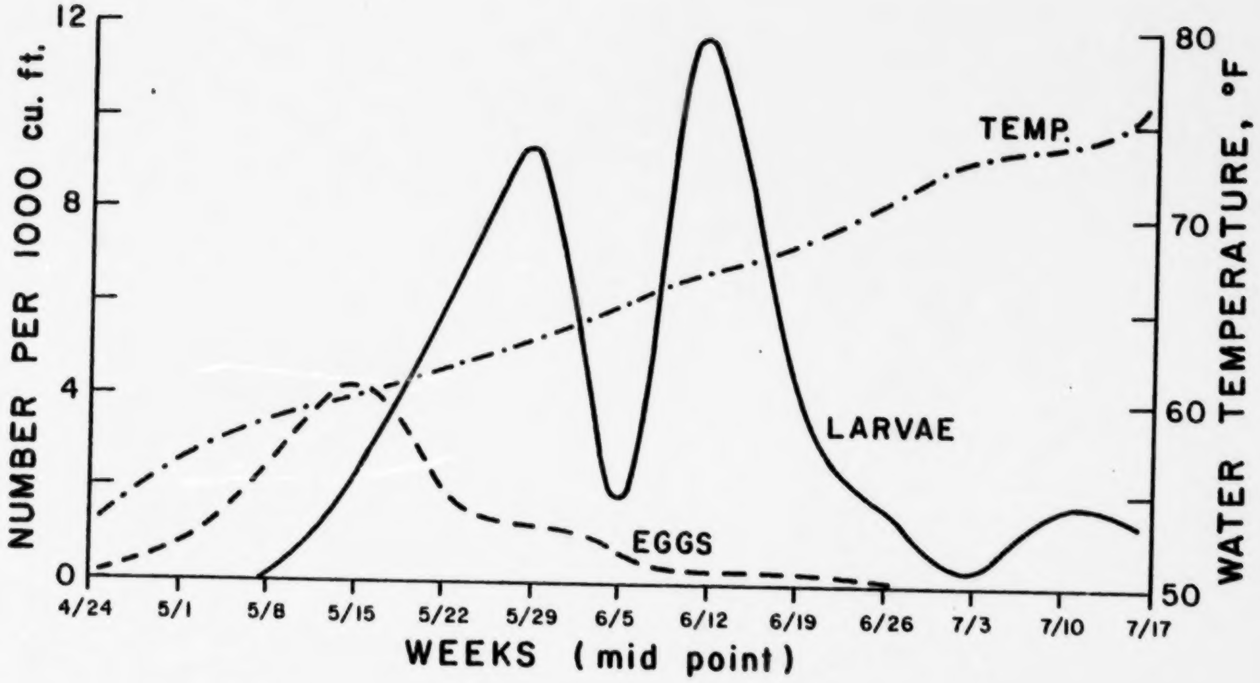


Figure 8. association of striped bass egg and larval abundance with water temperature in the Hudson River, New York, during 1968 (adapted from Carlson and McCann, 1969).

TABLE 9. ENVIRONMENTAL REQUIREMENTS OF STRIPED BASS EGGS

	ABIOTIC FACTORS	
	Survival Range	Optimum Conditions
Temperature	12-24°C	16-20°C
Salinity	0-15 o/oo	2-10 o/oo
Dissolved oxygen	>7% (3.3 mg/l @ 18°C)	air saturated
Light	no adverse effect	
Turbidity	0-1500 mg/l+	≤500 mg/l*
	BIOTIC FACTORS	
Diet	not applicable	
Density	50-75 per liter	
Predators	many in natural habitat	
Disease and Parasites	fungus	

+ clay and silt
* fine grain sediments

larvae at the higher temperatures and longer exposure times.

Striped bass egg survival is enhanced at low salinities. Salinities observed during spawnings were as high as 10 o/oo. Morgan and Rasin (1973) observed no significant effect of salinity from 0 to 8 o/oo on percent hatch or survival. Lal _et al_. (1977) reported that salinity of 3.4 o/oo enhanced survival. Optimal salinity for embryonic growth and survival has been found to be 0-1 o/oo at 18°C (Turner and Farley, 1971). Studies performed to determine the interaction of temperature and salinity effects on hatching were originally (1974) limited to three temperatures and six salinities. The percent survival (based on ten eggs per treatment) to hatching of the eggs stocked at 10-15 hours after fertilization (see Figure 5) was typically as shown in Table 10. In the next salinity-temperature interaction studies (1975) the number of temperatures was expanded to five and the salinities used were limited to the five at which survival was observed in the earlier experiments. The number of early eggs stocked in these experiments was 20 per treatment and the percent survival to hatching was typically as shown in Table 10. The information on abnormalities (Table 10) is given here but is discussed in more detail later in this section. The data presented represent the interaction effects of temperature and salinity on embryo survival. The broad range of survivals indicates good survival from 14 to 20°C with salinities of 0 to 10 o/oo. A narrower optimal temperature range of 18 to 20° is suggested by our data.

The minimum oxygen level for normal hatching has been determined as 4.9 to 5.0 ppm at 17-18°C (O'Malley and Boone, 1972; Turner and Farley, 1971). Hatching has been observed at 2.0 ppm (or 4% saturation) at 17°C, but the prolarvae were inactive and/or abnormal in development (O'Malley and Boone, 1972). To determine the interaction effects between temperature, salinity and dissolved oxygen approximately 100 eggs were stocked into one liter glass bottles of water at the stocking temperature (16-18°C) and 0 to 10 o/oo. After stocking into these well aerated bottles, they were transferred to 12, 16 or 20°C constant temperature baths and supplied with continuous dissolved oxygen at four levels of saturation - air saturated, 7% saturation, 5% saturation and 2% saturation. The levels were maintained by utilizing commercially available gas mixtures of percent oxygen with the balance as nitrogen. The percent survival, or the number hatched, alive and active, after 72 hours exposure is shown in Table 10. Our studies indicate that.the critical oxygen level for striped bass embryo development is primarily affected by temperature. Whereas there was some survival at the 2% saturation oxygen levels at 12°C, this was not evident at the higher temperatures and the larvae were very retarded in their development. Although the eggs hatched after exposure to 5% saturation levels at both salinities and most temperatures, these larvae were abnormally developed. Levels below 8% saturation would not, therefore, be considered optimal.

One observation (Albrecht, 1964) did not reveal any adverse effect of sunlight upon hatch and survival of embryos. All of our large scale cultures maintained in the field were held in full sunlight. We did not notice any difference in survivals of these eggs and subsamples used in experiments in our trailer-laboratory.

TABLE 10. PERCENT SURVIVAL TO HATCHING OF STRIPED BASS EGGS STOCKED AT VARIOUS (A) TEMPERATURE AND SALINITY AND (B) TEMPERATURE, SALINITY, AND DISSOLVED OXYGEN COMBINATIONS

A.

Salinity (o/oo)	Incubation Temperature (°C) 12	13	14	16	18	20
0	100	0*	90$^+$	70*, 91	100$^+$	60*, 80
5	68	70	95	90, -	100$^+$	90, 95
10	65	90	91	90, 93	100$^+$	70, 100$^+$
15	55	70	94	90, 67	100	80, 100
20	5	70	95	90, 53	86	70, 100$^+$

* The low survivals in freshwater (1974), in which the eggs are normally incubated in nature, is discussed in the abiotic factor section below.

+ Denotes abnormal or dead larva present but hatched.

B.

Salinity	Dissolved Oxygen (% saturation)	Incubation Temperature (°C) 12	16	20
0	100	77	83	86
	7	68	74	82
	5	61	0	66
	2	79 (unhatched)	0	0
10	100	84	75	90
	7	71	27	69
	5	83	19	0
	2	10	0	0

Using fine grain natural sediment from upper Chesapeake Bay, Auld and Schubel (1978) observed no significant effects on hatching of striped bass eggs in concentrations of 50, 100, or 500 mg/l. They reported significant reduction in hatching at 1000 mg/l concentrations of suspended sediments. Morgan et al. (1973) found significantly lower egg development in concentrations above 1500 mg/l of clay and silt from the Chesapeake-Delaware Canal.

Bowker et al. (1969) found pH ranges from 6.6 to 9.0 to be satisfactory for hatching.

Some of our studies included determining effects of ammonia and nitrate on embryo development. Using standard bioassay methods (APHA, 1965) for determining concentrations from stock solutions of ammonium chloride and sodium nitrate, tests were run on ten individual eggs per concentration. The percent survival to normal hatching for the ammonia concentrations at two temperatures (pH 6.8) was typically that presented in Table 11. The percent survival to normal hatching at the nitrate concentrations (pH 7.3) tested is also shown in Table 11. It appears that ammonia has little or no effect on hatching at 16°C, but generally reduces success by about half at 20°C. Nitrate concentrations of up to 1000 ppm should have little effect on hatching.

Exposure to shear levels (from laminar flow) of 350 dynes per cm^2 killed 36% of the eggs in one minute and 88% in four minutes (Morgan et al., 1976).

Survival of striped bass eggs to impingement on screens of 16 and 30 meshes per inch was 70% or better at water velocities less than 1.0 fps for four minutes exposure. Survival decreased sharply at higher velocities (Sazaki et al., 1972).

Results of egg exposure to changes in hydrostatic pressure, ranging from 2.0 psia (subatmospheric pressures) to 700 psig, are reported by Beck et al. (1975), and New York University, Institute of Environmental Medicine (1976). Later egg stages (close to hatching) were observed to be more sensitive than early egg stages, and decompression increased mortality.

Environmental factors affecting striped bass survival from spawning through embryo development have been summarized by Talbot (1966) and Dovel and Edmunds (1971). Additional comments specifically concerning Chesapeake Bay can be found in Mansueti (1961). The requirements discussed by these authors include water velocity, quality, turbidity, and temperatures.

Biotic Factors

There are four categories of biotic factors listed in Table 9 for embryos. However, for this life stage diet is not applicable.

Density can be considered minimal until the end of the stage when hatching begins. This is the time when oxygen demand increases. There is also an increase in the ammonia concentration with hatching. We have

TABLE 11. PERCENT SURVIVAL TO HATCHING OF STRIPED BASS EGGS EXPOSED TO VARIOUS CONCENTRATIONS OF (A) AMMONIA (NH_3) and (B) NITRATE (NO_3)

A.

Concentration (NH_3 ppm)	Incubation Temperature (°C) 16	20
Control	70	30
0.1	100	55
0.32	80	55
0.56	60	50
1.0	80	50
3.2	90	33
5.6	100	44
10.0	70	33

B.

Concentration (NO_3 ppm)	Incubation Temperature (°C) 18
Control	90
10	80
56	100
100	90
560	100
1000	70

measured the production in groups of 50 or 100 rinsed eggs at 15°C against blank replicates. The production ranged from 0.088 to 0.236 µg NH_3-N per egg (mean 0.157 µg; N = 7). This represents a release of 0.072 to 0.193 mg NH_3-N per 10,000 eggs on a live weight basis. Neither oxygen nor ammonia pose a problem to the embryos in their natural habitat, but they do become considerations in the more densely stocked culture environment.

The most prevalent disease and predator problem during the embryo stage was fungal and/or bacterial attack of the egg chorion. This was a factor contributing to freshwater mortalities in early temperature-salinity and other experiments during our studies using filtered river water. Dead eggs not only from our containers, but also from the river, were observed coated with continuous fungal hyphae strands extending through the chorion and into the yolk of living eggs. Unfortunately the fungi we observed contained no fruiting bodies making identification difficult beyond Saprolegnia sp. A series of test containers were treated with antibiotics to see if this treatment enhanced egg survival indicating reduction in fungal or bacterial activity. We use penicillin-streptomycin combination (50,000 IU/l and 50 mg/l), tetrocycline (6.25 mg/l), chloroamphenicol (50 mg/l), and sulmet (4 tbsp./gal.), in filtered river water plus a control of filtered river water. The dosages used were those found in the literature. (A very good report on the effects of antibiotics on survival of a marine fish is given in Struhsaker et al., 1973.) These containers were stocked with early eggs to observe the nature of hatching, and samples of the antibiotic treated and control river water were supplied to the Maryland Department of Health for total count testing. All of the eggs stocked into the chloromphenicol treated water developed abnormally. All of the eggs in the sulmet treated water hatched but died with fungus present. The eggs stocked into the control, tetrocycline, and penicillin-streptomycin treated containers all hatched normally. The total counts on the water samples from these treatments were 460, 28 and <3 MPM/100 ml, respectively. The penicillin-streptomycin combination was tested on netted eggs in filtered Nanticoke River water at 16 and 20°C and on newly fertilized eggs in Hudson River water at 15 and 18°C to determine its effect on egg survival. The percent survival in each group was greatly improved in the antibiotic treated water, as the example below shows.

TABLE 12. THE EFFECT OF TREATING FILTERED RIVER WATER WITH PENICILLIN-STREPTOMYCIN (50,000 IU/1-50 mg/1) ON THE PERCENT SURVIVAL AT HATCHING OF STRIPED BASS EGGS AT FOUR TEMPERATURES. (n = NUMBER STOCKED)

Treatment	Incubation Temperature (°C) 15	16	18	20
Penicillin-Streptomycin	70.2 (n=928)	97 (n=250)	62.5 (n=1041)	88 (n=257)
no treatment	9.0 (n=529)	92 (n=271)	7.3 (n=975)	69 (n=292)

Two antifungal agents were bioassayed using live eggs and also tested for their effectiveness using groups of dead eggs. Both series were done in triplicate. Untreated dead eggs quickly developed hyphal tufts and the effectiveness was judged by the presence or absence of hyphae on the dead eggs. Concentrations of 0.01 to 1.0 mg/l were tested for both Amphotericin B and malachite green for 24 hours at 16°C. The Amphotericin B was toxic to eggs at concentrations lower than that at which it was effective in controlling fungus on dead eggs (1.0 mg/l). The malachite green was effective at the low concentrations with no evidence of fungal activity at 0.01 mg/l and no egg mortality at concentrations lower than 0.5 mg/l.

CULTURE METHODOLOGY

Capture Methods

Fertilized striped bass eggs can be obtained from natural or artificial spawnings. Producing artificially spawned eggs is discussed in the section dealing with adults. In addition to obtaining eggs from artificially spawned females, live eggs can be collected during the spawning season using a regular 1/2 meter plankton net. McCoy (1959) recommended a 500 micron mesh as optimal for collecting striped bass eggs. Although such tows yielded hundreds of eggs which we used in our laboratory work, the mesh became rapidly clogged.

We used a 1 X 2 meter 945 micron neuston net that was modified to include floatation gear on the heavy steel frame to make it float just below the surface. The net was easily fished by securing its bridle to a bridge pier, pier or docked vessel and allowing it to stream in the tidal currents. The large filter area to mouth opening ratio provided by the net's 23 foot length allowed large volumes of water to be filtered. Clogging was never a problem even after 4 to 6 hours of fishing. Some large debris was inevitably collected. A large mesh preventer net over the mouth of the net reduces this problem.

Tows made near the spawning peak yielded tens of liters of nearly solid striped bass eggs. The egg catch from an early tow filled a plastic garbage can. Eggs were separated from plant debris by raising the salinity to 10 o/oo which allowed the eggs to float. Eggs were then decanted to hatchery containers. There were always some other species present. Night tows frequently yielded large numbers of elver eels and juvenile croakers which were difficult to remove. We thus avoided night tows where possible.

We recommend this approach to secure large numbers of fertilized eggs easily without having to capture and hold females and males. These net caught eggs, moreover, represent a diverse genetic stock not available from the progeny of a given mating under artificial spawning conditions. It is a simple reliable method.

Post-capture Handling

Eggs secured from plankton tows should be separated from the rest of the tow as described above before packing for transportation or stocking into rearing containers. Eggs secured from artificial spawnings should be stocked into rearing containers and allowed to fully water harden. They should not be transported until at least 12-24 hours after fertilization to avoid dead egg accumulations due to poor fertilization. We recommend using penicillin and streptomycin (50,000 IU/l and 50 mg/l) in the culture water receiving the eggs. This antibiotic concentration should be applied once only when the eggs are stocked. If heavy fungus infestation is visible a flush treatment of malachite green at 0.1-0.05 mg/l is recommended.

Transportation

Egg transportation can be successfully undertaken using methods described by Bayless (1972) and Texas Instruments (1977c) for larvae. This involves packing about 15-20,000 eggs per liter into plastic bags lining a styrofoam fish shipping box. In this way as many as 200,000 eggs can be shipped in a container about half filled with water and eggs. Before the bag is secured oxygen is bubbled into the water and allowed to fill the space over the water in the bag. A dose of penicillin-streptomycin can be added for the volume of shipping water. This is recommended for shipments over long distances.

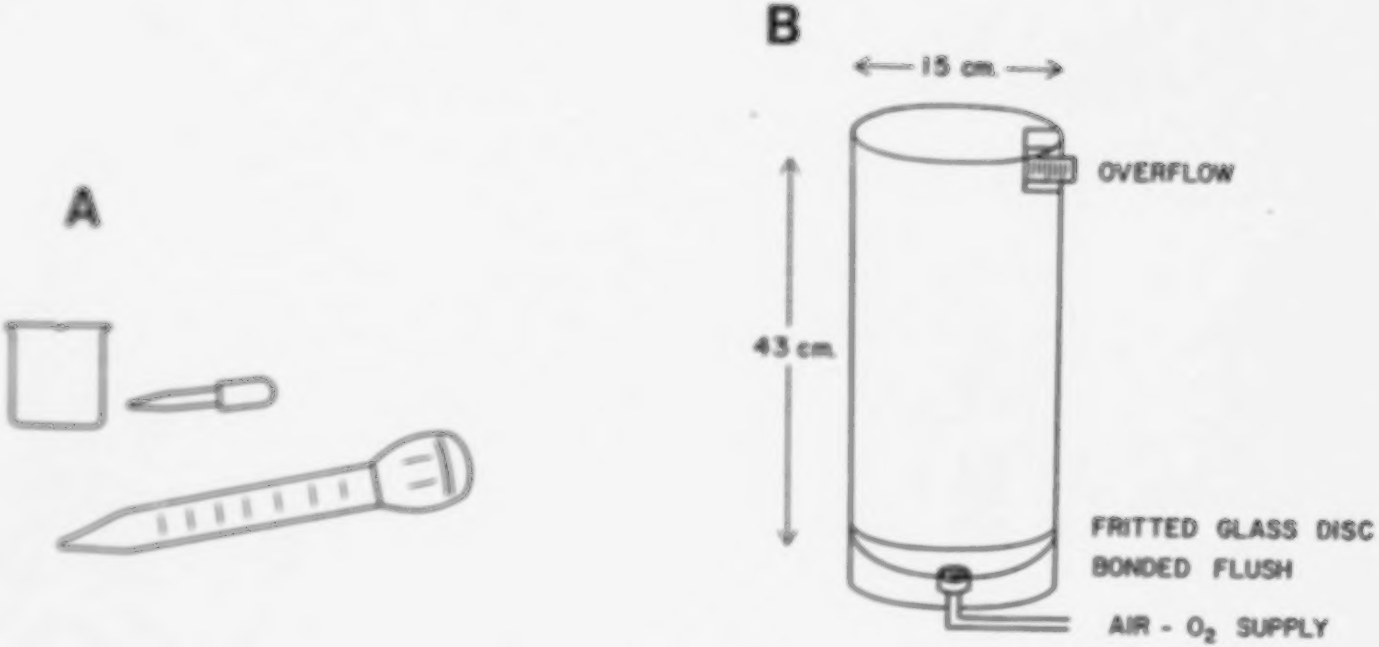
Handling Procedures

Eggs should be handled in water whenever possible. They can be easily dipped, siphoned or pipetted with a wide bore tube. Several useful egg handling tools are illustrated in Figure 9A. Dip nets are not recommended for live egg handling. Dead eggs, which are opaque and float to the surface, may be skimmed from the rearing container using a net or beaker. Individual eggs can be easily counted using a wide bore pipette. A rough approximation can be made using volume displacement in rearing water of a known number of eggs. After 'calibrating,' this measure is then repeatedly estimated. The most accurate counts, however, will be those of individual eggs.

Maintenance Procedures

Culture Vessels--

A number of culture vessels have been used in experimental work involving relatively small numbers of eggs per treatment. Schubel (1974) used a hatching box made out of a PVC frame covered with nylon screen. Miller (1977) used a hatching basket similar to this but made of acrylic and screened with 505 μ Nitex. Turner and Farley (1971) used a simpler egg container made from a section of 2 1/2 inch PVC pipe, one end of which was covered with stainless steel bottling cloth. Eldridge et al. (1977) used glass hatching jars, while Rogers et al. (1977) used glass beakers. The Moncks Corner Hatchery (Bayless, 1972) relies on acrylic McDonald hatching jars in a flow-through system. These are similar to the hatching jar illustrated in Figure 9B which can be used in a flow-through or static system.



A
B
15 cm.
43 cm.
OVERFLOW
FRITTED GLASS DISC
BONDED FLUSH
AIR - O_2 SUPPLY

Figure 9. A. Suggested tools for handling eggs and larvae of striped bass so that the animals remain in the water.

B. Suggested striped bass egg hatching container modified from a McDonald hatching jar. The dimensions indicated are those of the original hatching jar. However, the container can be enlarged to accomodate the needs of a large scale rearing system.

For large volume egg maintenance we used (in Rhode Island) 55 gallon polyethylene drums filled with dechlorinated tap water and agitated with a strong stream of air or a gentle stream of pure oxygen. Either of these provided enough agitation to keep the eggs in suspension and maintain an adequate dissolved oxygen level. The water temperature of the rearing tank and transporting container should be about equal at stocking.

Stocking Density--

Bonn *et al*. (1976) recommend stocking fertilized eggs at the rate of 100,000 per hatching jar. This is approximately 1777 per liter. Literature reports of experiments under static conditions on egg stages report stocking densities from 20 (Albrecht, 1964) to 100-200 (Rogers, 1978) per liter. At the rate of 100 eggs per liter, 20,000 eggs could be easily handled in the static 55 gallon drums described above. In fact, we estimated that twice to three times this density were easily held in these tanks through hatching.

Maintaining Water Quality--

Maintaining water quality through the embryo stage in any culture system is relatively easy for two reasons. First, the length of the period is very short (see Table 7) and second, the physiological demands during the stage are minimal. Care of the egg cultures should include removing any dead eggs at least daily. Toward the end of the stage care should be taken not to remove any newly hatched larvae which are also at the surface with the dead eggs. Oxygen levels should be monitored to ensure adequate saturation. If the antibiotic dosage recommended (see biotic factors above) is followed, no water quality problems associated with fungal or bacterial infections should be prevalent. If this dosage is not utilized and an outbreak of fungus is observed, a malachite green flush is recommended.

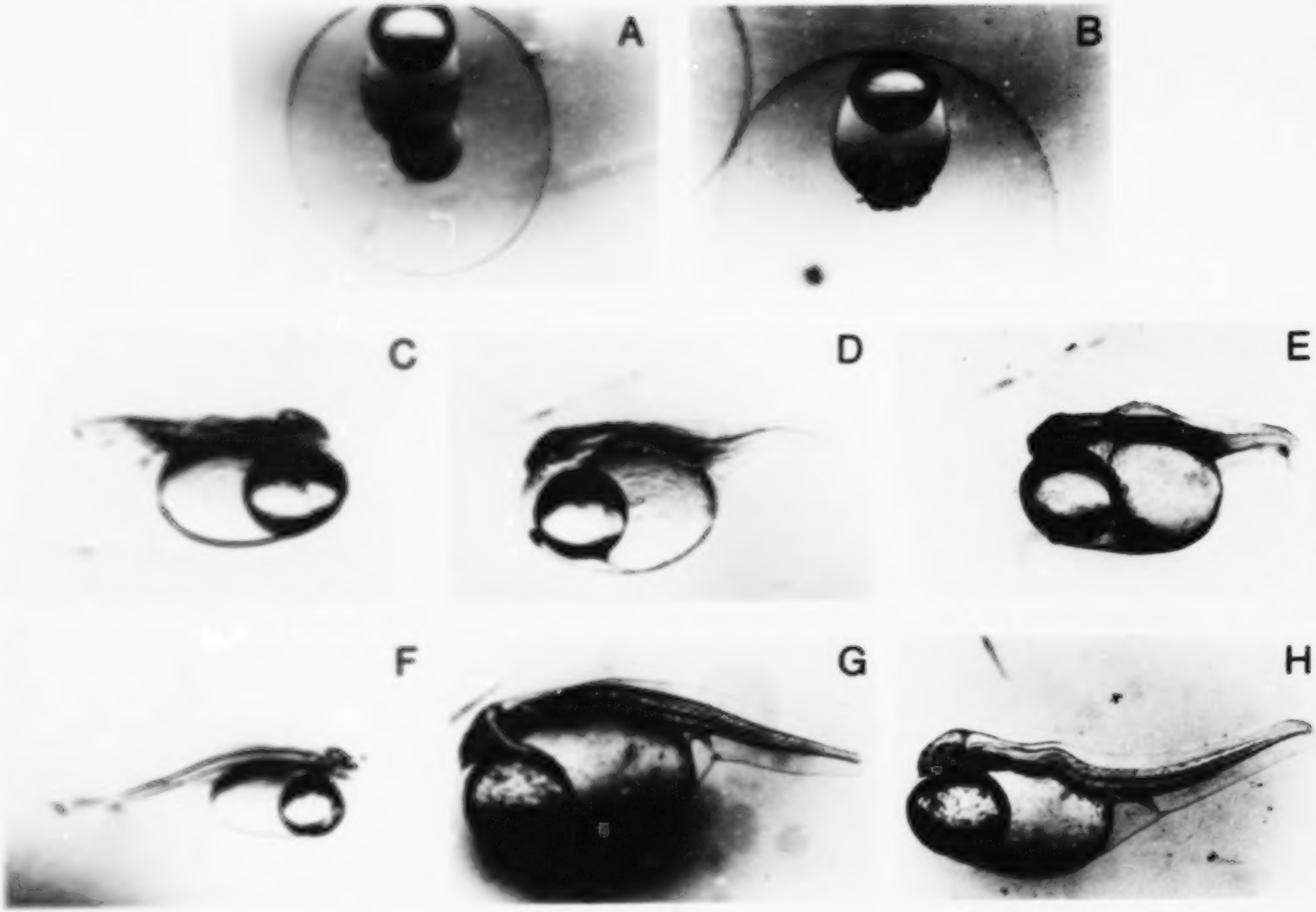
Diet--

This is not relevant for this stage, which utilizes the endogenous energy supplied in the yolk.

Normal Conditions and Physiological State

Normal development through the embryo stage has been presented in Figures 5 and 6. Abnormal conditions were reported by Worth (1910), Scofield and Coleman (1910), Mansueti (1958), O'Malley and Boone (1972), and Koo and Johnston (1978). Many of the abnormalities they reported we also observed. Some of the more frequent ones are shown in Figure 10 from our observations. One which occurs under a variety of conditions is a premature loss of the chorion, or early hatching. An embryo which has lost its chorion early will usually not be as active as is a normal newly hatched larva (described in the next section). Most of the embryos we observed in this condition grew into normal larvae. The antibiotic treatments appeared to reduce this phenomenon, especially among the eggs in our large scale cultures maintained in filtered river water while in the field.

Figure 10. Some of the more common abnormalities of striped bass embryos before and just after hatching: A,B - lethal abnormalities occurring in eggs soon after fertilization; C,D,E - examples of living larvae showing arrested body development of the sort occurring in association with hypoxic stress during egg development (generally lethal); F,G,H - yolk sac and skeletal abnormalities in newly hatched larvae (generally not immediately lethal)



68-12

SECTION 9

RECOMMENDED CULTURE METHODS AND BIONOMICS: LARVA

DESCRIPTION OF STAGE

The larval stage of the striped bass can be divided into prolarval (hatching to yolk absorption and feeding) and post (yolk sac) larval (yolk absorption and feeding to metamorphosis) periods. Development during the prolarval and postlarval periods is shown in Figure 11, where (d) is yolk absorption. A more detailed description of the progression from hatching through metamorphosis is found in Table 13. The rate of development depends on the temperature experienced as indicated in Figure 12. The duration of these stages have been estimated to range from 3.8 to 10 days and 22 to 76 days, respectively, depending on temperature (Lawler *et al.*, 1974; Rogers *et al.*, 1977; USNRC, 1975) and nutritional state (Rogers *et al.*, 1977).

Measurements made at hatching (Figure 11a) on larvae from eggs held at 15, 18, 21 and 24°C are shown in Figure 13. At hatching the yolk sac and oil glouble are the most conspicuous features of a larva. As development progresses (Figure 11), the yolk material is used while the oil glouble remains essentially unchanged. Figure 14 shows measurements on larvae at yolk absorption from four rearing temperatures. A comparison of Figures 13 and 14 shows that as the yolk disappears the oil volume remains about the same, while the larva grows in both embryo length and dry weight (larval tissue less yolk and oil in both cases). Larvae begin feeding during the later part of the yolk absorption period (Figure 11 c-d). Swim bladder inflation (Figure 11e) normally occurs about 5 to 7 days after hatching (Bulak, 1976; Doroshev and Cornacchia, 1979), although Bulak (1976) observed that 60 day old larvae with previously uninflated gas bladders were able to initiate filling. Following yolk absorption with the continuation of successful feeding, the larva utilizes the lipid energy in the oil glouble and development proceeds as indicated in Figure 11e-h. However, if the larva does not feed successfully or is starved, the energy in the oil glouble is conserved (Dergaleva and Shatunovskiy, 1977; Eldridge *et al.*, 1977; Rogers and Westin, 1979). Thus larvae dying of starvation will look essentially like those at yolk absorption with the oil glouble conspicuously present (Figure 11c-d). Retention of the oil can be seen in the percent nitrogen and carbon composition of larvae measured at yolk absorption and of those measured seven days after yolk absorption. These determinations are summarized from Rogers (1978) in Table 14 and include the carbon-nitrogen changes among feeding larvae as well.

Some of the increase in larval length during the prolarval period is probably due to hydration as indicated in Figure 13. The length-dry weight

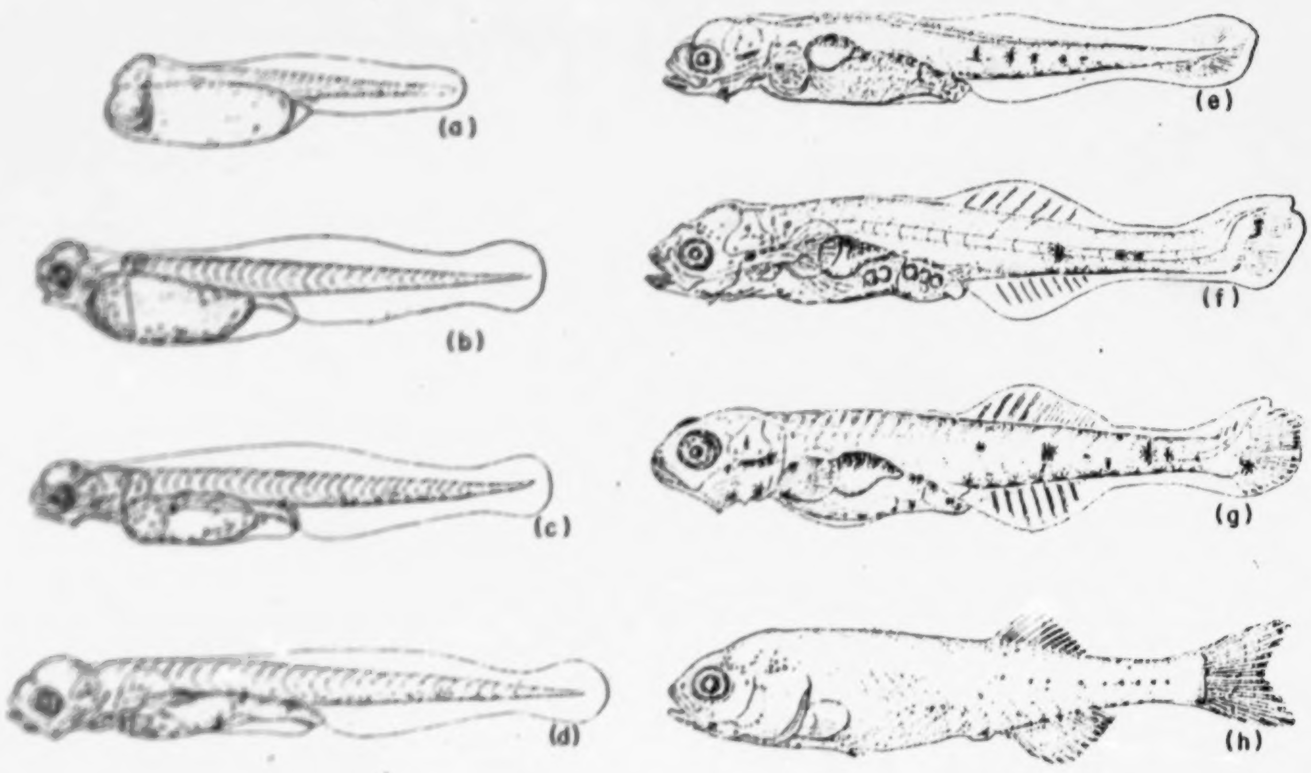


Figure 11. Developmental stages (after Manuseti, 1958) of striped bass larvae to metamorphosis. Refer to Table 13 for further description of stages.

TABLE 13. DEVELOPMENTAL STAGES OF STRIPED BASS, REARED AT ABOUT 17°C, UNLESS OTHERWISE STATED, THROUGH TRANSFORMATION

Age	Length mm TL[a]	Characteristics
25.8 hours after fertilization (4)[b]	3.25-4.06	Hatching completed for eggs at 24°C. (a)[c]
36-48 hours after fertilization (2)	2.5-3.7	Hatching occurs.(a)
51.8 hours after fertilization (4)	3.25-4.71	Hatching completed for eggs at 18°C. (a)
1st day after hatching (4)	3.58-5.12	Eyes almost fully pigmented; pigmented ventrally; one-third yolk reabsorbed at 24°C.
	4.23-5.20	Eyes only partially pigmented; yolk slightly reabsorbed at 18°C.
2-5th day after hatching (1,2)	4.5-5.2	Yolk sac partly absorbed, eyes pigmented yellow, black & orange, differentiation of jaws and digestive tract begun, pectoral buds formed fan-like fin, 21-23 myotomes. (b)
3rd day after hatching (3)	5.2	Eyes pigmented, jaws developing, pectoral fins become differentiated.
(4)	4.71-6.23	Eyes pigmented; mouth parts moving; pigmented ventrally jaw to oil; yolk three-fourths reabsorbed; pectoral buds present at 24°C.
	5.04-5.77	Eyes pigmented; gut differentiated; ventrally pigmented; pectoral buds visible at 18°C.
4th day after hatching (3)	5.8	Small chromatophores along ventral edge of entire yolk sac.
(4)	5.5-7.5 (live)	Yolk absorbed at 24°C.
5th day after hatching (1)	5.5-5.8	One-third yolk reabsorbed, commencement of intestinal peristalsis, 23-24 myotomes. Swimming pelagically(c).
6th day after hatching (3)	6.0	Oil glouble and yolk nearly absorbed, pigmentation ventrally.(c)
6th-7th day after hatching (4)	5.5-7.5 (live)	Yolk absorbed at 18°C.(d)
8th day after hatching (1)	5.8-6.5	Teeth on jaws, orange pigment in caudal (heterocercal) area, differentiation of stomach, three-fourths yolk reabsorbed, 25 myotomes. Transition to active pelagic feeding.(d)
(3)	6-9	Second dorsal and anal slightly differentiated well-developed mouth parts. (d)
10-15th day after hatching (2)	7.5	Yolk sac fully absorbed and no oil glouble visible, pectorals only fins visible, teeth visible, generally pigmented on body. (e)

(continued)

TABLE 13 (continued)

Age	Length mm TL[a]	Characteristics
10th day after hatching (3)	9.0	Pectorals only fins developed, ready for food.
15th day after hatching (1)	10-12.5	Division of fin fold into three divisions, complete reabsorption of oil glouble, single-chamber gas bladder filled with air. Feeding on plankton. (e)
18th day after hatching (3)	13.0	Dorsal and anal fin rays well differentiated and rudimentary spines observed. (f)
20-30th day after hatching (1,2)	10, 12-16	Differentiation of rays in caudal, anal and dorsal fins. First dorsal elements and pelvic fins absent, myotomes correlated with number of vertebrae. (g)
30 days after hatching (4)	13.1-15.4	Metamorphosis at 24°C.
30-40th day after hatching (2)	15 (stunted)	Soft dorsal, anal and caudal (homocercal) fins well differentiated, spinous and pelvic fins not well developed and well ossified, no stripes visible yet. Initial formation of lateral-line scales (Murawski, 1958). (h)
40 days after hatching (4)	11.9-20.4	Metamorphosis at 18°C
40-50th day after hatching (1)	22-35	Differentiation of rays in first dorsal and pectoral fins. Full complement of lateral-line scales by 30mm (Murawski, 1958).
50-70th day after hatching (1)	35-45	Scales
(2)	20	Scales observed for first time, fins except larval pelvic in various stages toward full meristic count, pigmentation stronger.
60-80th day after hatching (2)	25	Covered with scales, 3 anal spines and full complement of meristic characters, body covered with melanopores.
80-90th day after hatching (1)	50-60	Appearance of longitudinal stripes.
70-100th day after hatching (2)	30	Meristic counts complete except for pectoral fin rays, body pigmentated.
3-4 weeks after hatching (3)	36	Fully developed fins and rays, pigmentation of black dots.

[a] Total length measured on preserved samples unless otherwise stated.

[b] Numbers in parenthesis refer to source, i.e., (1) Doroshev (1970); (2) Mansueti (1958) ; (3) Pearson (1938); and (4) Rogers et al. (1977).

[c] Letters in parenthesis refer to Figure 11.

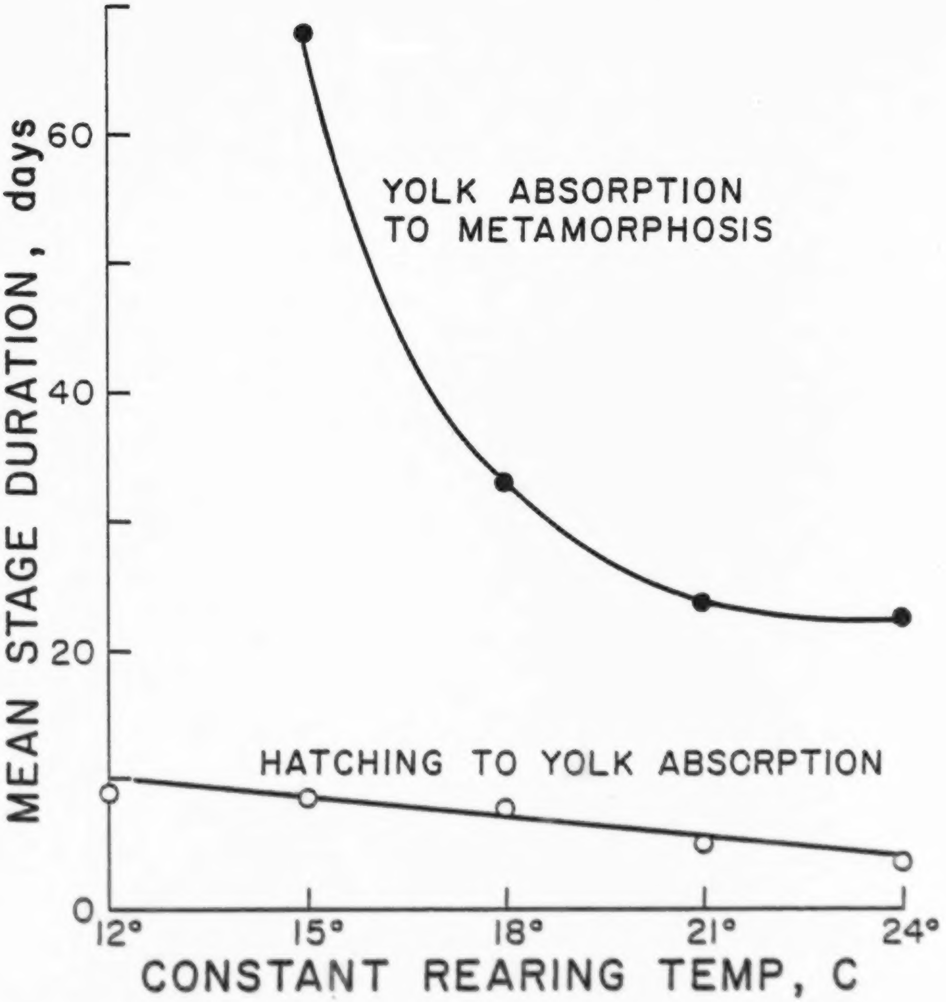


Figure 12. The effect of rearing temperature on the duration of the yolk sac and larval stages of striped bass. Each point represents the mean of at least three stage duration observations at each temperature treatment. (Rogers *et al.*, 1977)

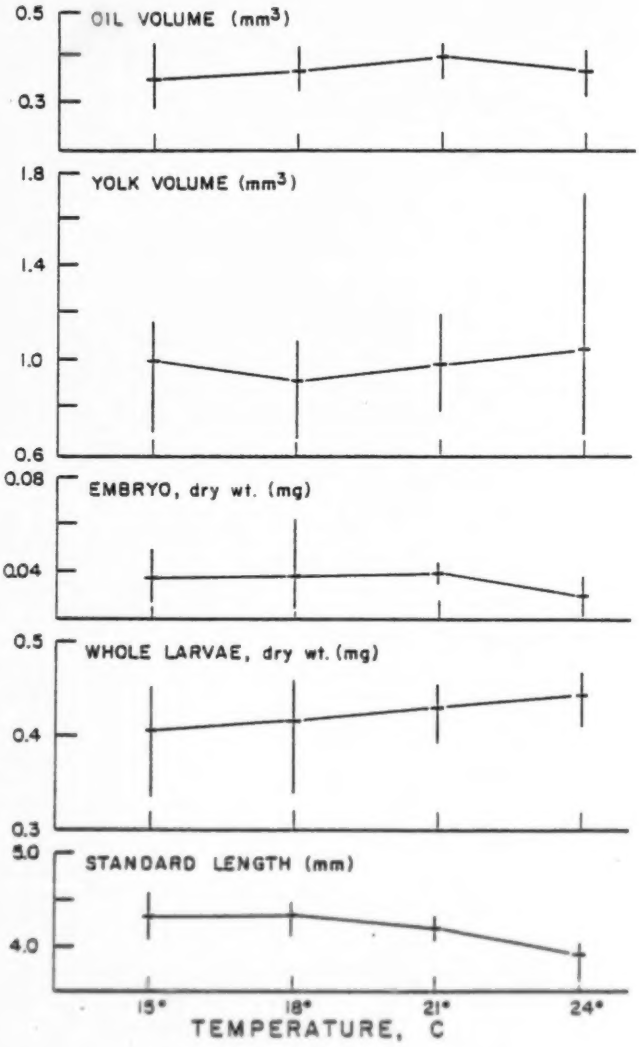


Figure 13. Measurements made on New York 1977 newly hatched striped bass prolarvae after incubation at four temperatures. Each measurement is of ten individuals. (Rogers, 1978)

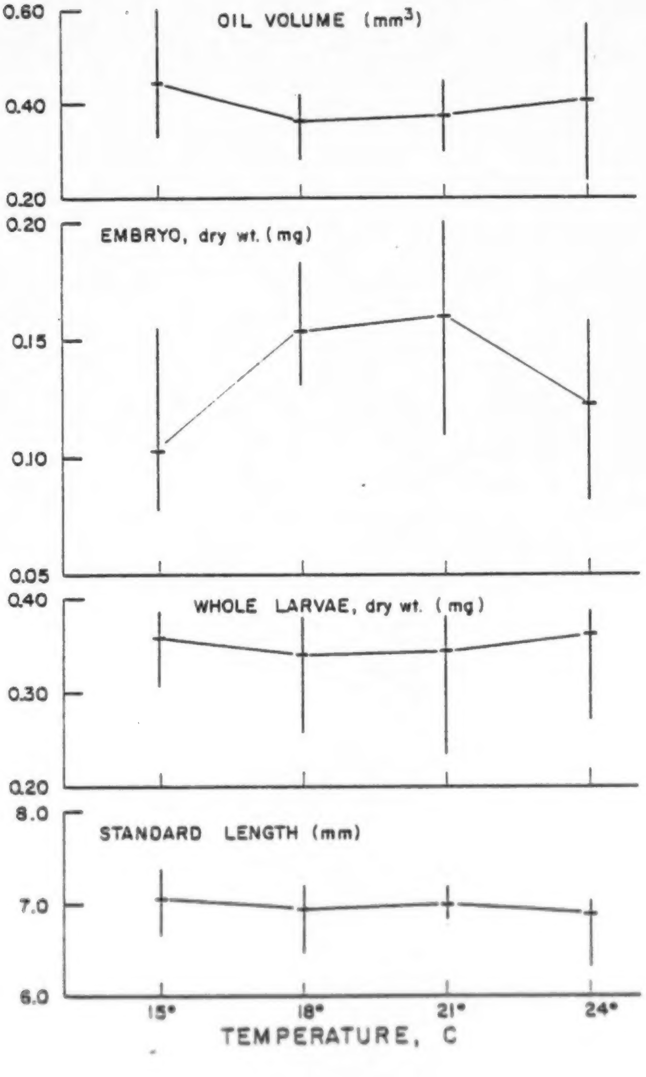


Figure 14. Measurements made on New York 1977 striped bass prolarvae at yolk absorption after incubation and maintenance at four temperatures. Each measurement is of ten individuals. (Rogers, 1978)

TABLE 14 . AVERAGE PERCENT COMPOSITION (CARBON AND NITROGEN) OF STRIPED BASS PROLARVAE, LARVAE AT YOLK ABSORPTION, AND FED AND STARVED POSTLARVAE REARED AT FOUR TEMPERATURES

Larval Stage	Sample	% of composition of sample		Ratio
		nitrogen	carbon	C:N
Yolk sac larvae	5 replicates of 4 larvae each	4.60	57.06	12.40
Larvae at yolk absorption	5 replicates of 5 larvae each	6.04	60.40	10.00
Larvae, 7 days after yolk absorption:				
24° fed	groups of 5 larvae each	8.20	48.10	5.86
starved		5.18	61.60	11.89
21° fed		8.48	46.68	5.50
starved		6.25	51.45	8.23
18° fed		8.40	48.36	5.76
starved		5.84	53.30	9.13
15° fed		6.35	53.97	8.49
starved		5.63	51.86	9.21

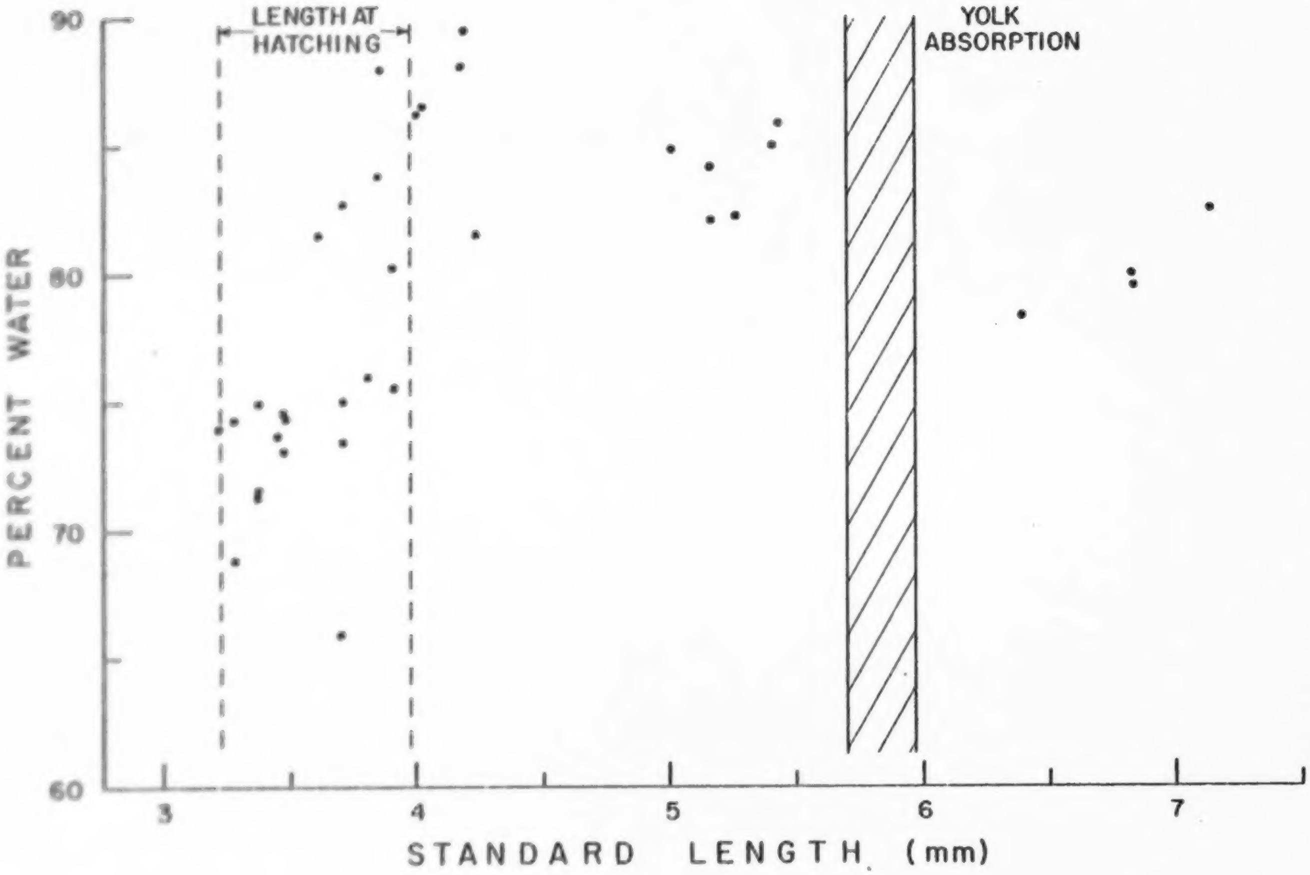


Figure 15. Percent water content of striped bass prolarvae. (Rogers *et al.*, 1977)

relationship for healthy larvae 3 to 25 mm standard length (SL) is given by the equation:

$$\log_{10} \text{dry weight (mg)} = \log_{10} \text{SL(mm)}\ 2.952 - 2.707$$

($n = 185$; $r = 0.932$) from our measurements.

NATURAL HABITAT

Larvae are planktonic, drifting in a head-up position due to the buoyancy of the oil-yolk sac at hatching. As the larvae develop toward yolk absorption (Figure 11) their swimming movements become less irregular. Postlarvae are able to resist current movements on the spawning grounds. As metamorphosis approaches, their movements are very well directed.

Larval distribution in relation to various temperature, conductivity (salinity) and dissolved oxygen levels are represented in Figure 16 (prolarvae) and Figure 17 (postlarvae) in the Hudson River, and in Figure 18 for the Potomac River. Larvae are generally found on the spawning grounds at temperatures from 15 or 16 to 22 or 23°C, salinities of 0 to 6 o/oo (4 o/oo = 7 mS5 cm^{-2} conductivity), and dissolved oxygen levels of 7 to 10 mg/l (or >78% saturation at 20°C). Depending on the temperature (Figure 12), larvae are present in the river areas about 4 to 6 weeks after the last spawnings.

ENVIRONMENTAL REQUIREMENTS

The major abiotic and biotic factors discussed below are summarized in Table 15 for the prolarval and postlarval periods.

Abiotic Factors

The optimum temperature for larval growth is 15-22°C (Davies, 1970; Bogdanov et al., 1967) and 15.6-18.3°C (Bayless, 1972). Rogers et al. (1977) proposed a physiological growth optimum of 18-21°C for the yolk absorption period. They found that growth rates among post-yolk sac larval striped bass were highest at 21 and 24°C. Morgan and Rasin (1973) observed minimal larval lengths at 13.5 and 16°C and maximal larval lengths at 21.5°C in their studies. Kelly and Chadwick (1971) reported the 48 hour LD50 for striped bass 5 to 38 mm to be from 28.9 to 32.8°C. The variation within this range was not related to either acclimation temperature or fish size. Carter et al. (1979) calculated the thermal doses needed to produce mortality of 10% of experimental post yolk sac larvae as a function of temperature from thermal resistance data collected by Ecological Analysts in experiments on the Hudson River. These calculations suggested that 10% of the larvae would suffer instantaneous death at 35°C regardless of their acclimation temperature (15, 20.5, 22, 23, or 23.5°C). Their calculations indicated incipent lethal levels of 23.6, 29.0, and 31.6°C for the larvae acclimated to 15, 20, and 23°C, respectively. Reduced food catching ability was

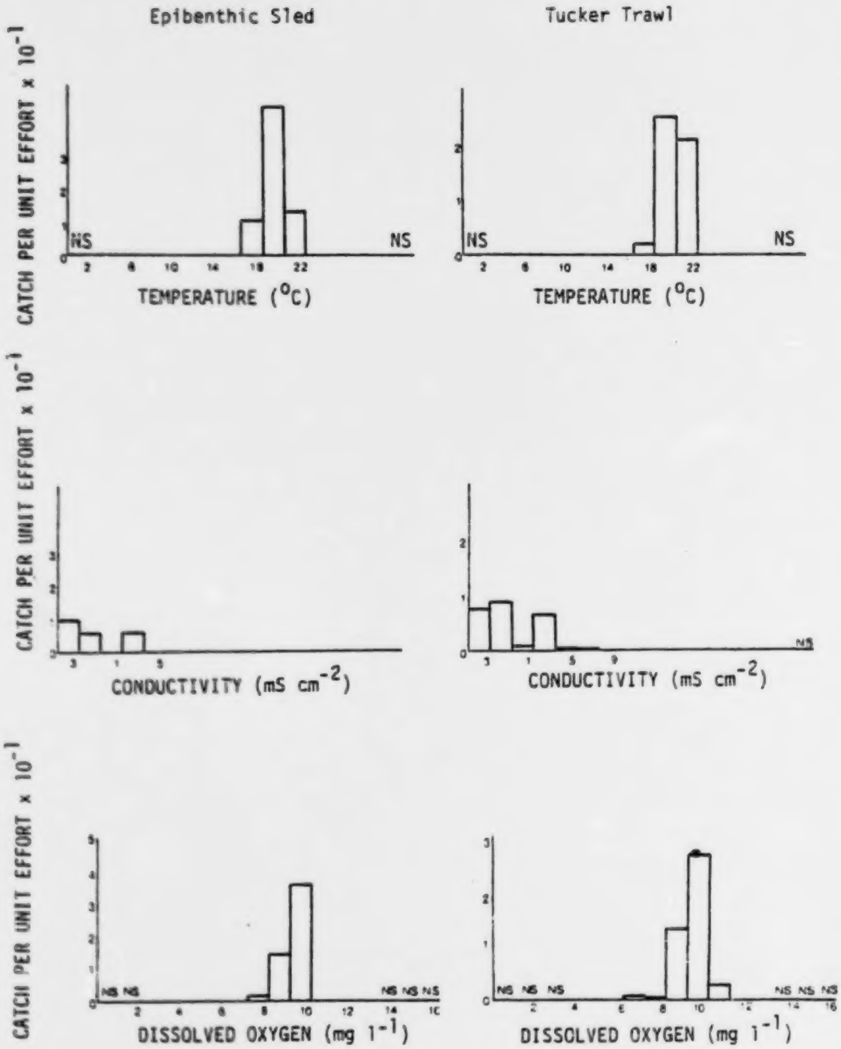


Figure 16. Catch per unit effort of striped bass yolk sac larvae collected by epibenthic sled and tucker trawl at various temperature, conductivity and dissolved oxygen concentrations in the Hudson River, New York (RM 14-140; km 23-227) during 1975. (adapted from McFadden, 1977a)

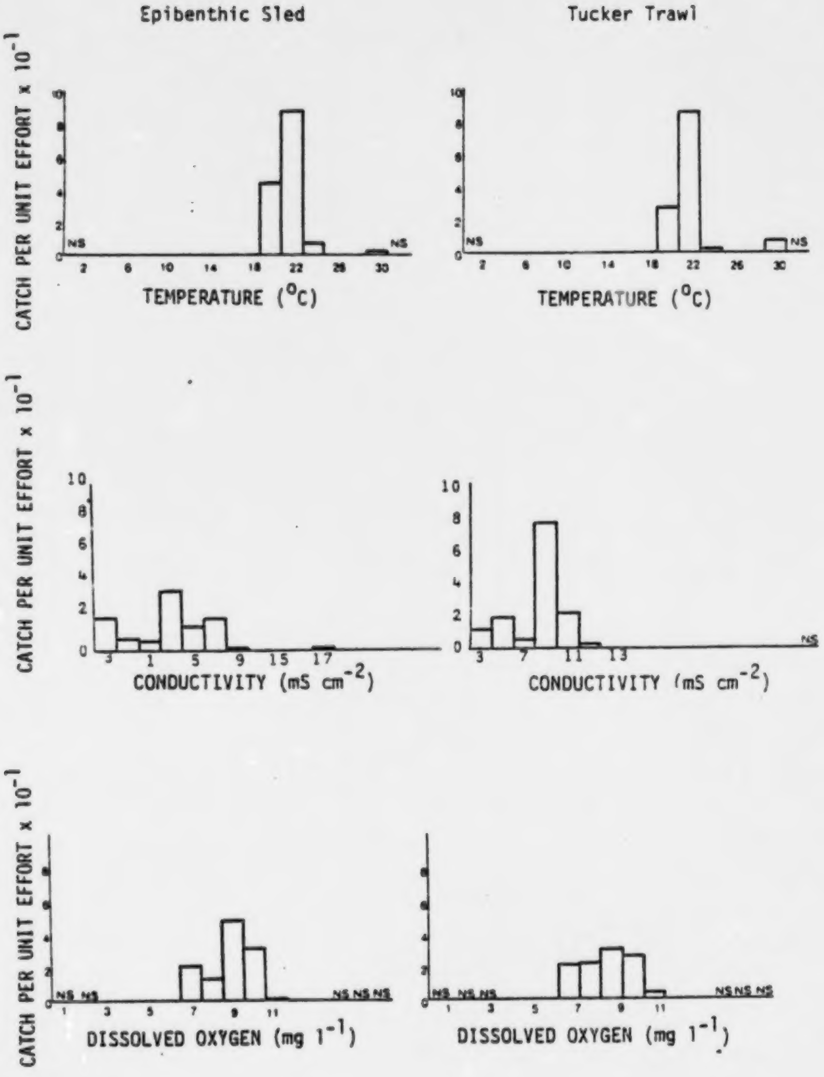


Figure 17. Catch per unit effort of striped bass post yolk sac larvae collected by epibenthic sled and tucker trawl at various temperature, conductivity and dissolved oxygen concentrations in the Hudson River, New York (RM 14-140; km 23-227) during 1975. (adapted from McFadden, 1977a)

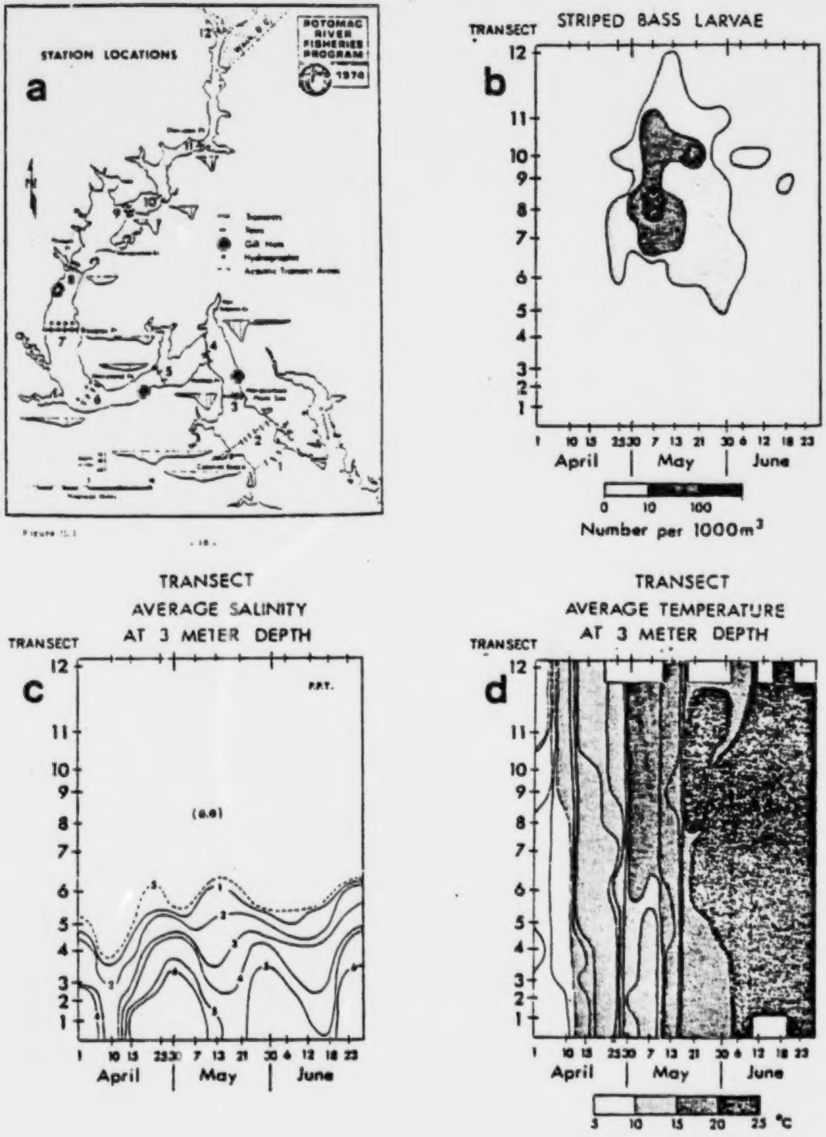


Figure 18. Larval density (b) in the Potomac River, Maryland (a), during 1974 over the salinity (c) and temperatures (d) reported. (adapted from Polgar *et al.*, 1975)

TABLE 15. ENVIRONMENTAL REQUIREMENTS OF LARVAL STRIPED BASS

ABIOTIC FACTORS

	Survival Range		Optimum Conditions	
	Prolarva	Postlarva	Prolarva	Postlarva
Temperature	12-27°C	10-27°C	16-21°C	18-22°C
Salinity	0-15 o/oo	5-25 o/oo	5-15 o/oo	10-20 o/oo
Dissolved Oxygen	>7% (3.3 mg/l @ 18°C)	>5% (2.4 mg/l @ 18°C)	air saturated	
Light	no adverse effect		natural photoperiod	
Turbidity	1-1000 mg/l[+]		≤ 100 mg/l[+]	

BIOTIC FACTORS

	Prolarva	Postlarva
Diet	not applicable	minimum of 1000-2000 nauplii/liter twice daily, or 15-20 % of body dry wt
Density	50-25 per liter	30-10 per liter
Predators	many in natural habitat	cannibalistic; many in natural habitat
Diseases and Parasites	for summary see Table 24	

[+] fine grain sediments

observed among postlarvae at 7.8°C (Hughes, 1967).

Good survival and growth during larval development were found at 3.5-14 o/oo salinity by Bayless (1972). Davies (1973) calculated optimal rearing conditions for yolk-sac larvae, based on observed survival under 15 different combinations of temperature, pH and total dissolved solids, of 17.6°C, pH 7.5, and total dissolved solids 185.7 mg/l NaCl. Lal et al. (1977) transferred prolarvae to various salinities at 18.5°C and reported the best survivals at 10% sea water (about 3.4 o/oo). Otwell and Merriner (1975) observed greater than 80% survival of bass larvae during seven days subsequent to direct transfer from their rearing facility into temperatures of 18 and 24°C and salinities of 4 or 12 o/oo.

In our 1974 and 1975 field studies we investigated the survival of prolarvae to various salinity and temperature combinations. After 24 hour exposure of two day old prolarvae, 90-100% survival was observed at temperatures of 13 and 16°C and salinities of 0, 5, 10, 15, and 20 o/oo. At 20°C, prolarvae survived well (95-100%) at salinities of 5, 10, and 15 o/oo. There appeared to be some temperature-salinity interactions, but prolarvae survived 15 o/oo salinity well. Similar experiments with postlarvae (Figure 11e) indicated good survival (95-100%) after five days exposure at salinities of 10, 15, 20 and 25 o/oo at 14 and 18°C. This indicates that older postlarvae can easily adjust to full sea water, however, others suggest that the best time to introduce larvae to sea water is just after metamorphosis (Lal et al., 1977).

A critical oxygen level of 1.65 mg/l and a suitable level of 5-6 mg/l dissolved oxygen have been reported for larval striped bass (Bogdanov et al., 1967). Turner and Farley (1971) reported holding larvae hatched from eggs exposed to 4 mg/l dissolved oxygen for varying periods (0 to 30 hours) for six days after hatching. They observed that the longer the eggs were exposed to low oxygen conditions, the lower the percent survival of larvae after six days. Bulak (1976) observed that reduced oxygen levels might adversely affect normal swim bladder inflation. Doroshev and Cornacchia (1979) observed that strong aeration seemed to enhance normal inflation. We investigated the interaction between temperature and dissolved oxygen levels on larval survival at 5 o/oo salinity. The percent oxygen was maintained using oxygen-nitrogen gas mixtures. The survival after a 24 hour exposure period was usually 100% at the air saturated and 10% saturated oxygen levels at the temperatures tested (13, 16, 18, 20, 21) for prolarvae. However, the 7% saturation levels (i.e. 3.3 mg/l @ 18°C) showed very reduced survival (5 to 85%) over the temperature range. There was no survival of prolarvae at any of the 5% saturation levels. The postlarvae, however, showed 90-100% survival at the two temperatures tested (18 and 21°C) for all dissolved oxygen levels from 5% saturation (2.4 mg/l at 18°C) to air saturation.

McHugh and Heidinger (1978) reported observations of light shock on three age groups of larvae. The youngest group (5-9 days old) showed the most active response to light shock (1238 lux) of an hour following three hours of dark. They dove for the bottom and swam rapidly for about two minutes. Larvae 11-23 days old responded only slightly to the light shock, while larvae 15-33 days old showed no activity or response. In 1977, they reported no significant difference in egestion time between larvae (9-19 days old) held in light and in darkness. Braid (1977) observed no difference in behavior of larvae exposed to light of various wavelengths.

Morgan et al. (1973) reported an LD50 for 2-day exposure of larvae to 3411 mg/l of clay and silt from the Chesapeake-Delaware Canal. Auld and Schubel (1978) reported exposure of yolk sac larvae to concentrations of natural fine-grained suspended sediments less than 100 mg/l did not significantly affect survival for periods up to 72 hours. Survival rates, however, decreased for larvae exposed to 500 and 1000 mg/l concentrations. The toxicity of several chemicals to larvae are presented in Section 12.

Bogdanov et al. (1967) reported pH of 7.5 as favorable for larvae reared in soft water. Bonn (1970) determined that pH of 10 was the upper lethal.

Studies we performed to determine the tolerance of larvae to nitrogenous compounds indicated that prolarvae were more susceptible to the effects of ammonia concentrations (standard dilutions of ammonium chloride stock) in freshwater. Percent survival after 48 hours exposure to the temperature, salinity and pH regimes tested is shown below based on the stocking of 20 individuals per concentration per test.

TABLE 16. PERCENT SURVIVAL OF STRIPED BASS PROLARVAE AFTER 48 HOURS EXPOSURE TO VARIOUS AMMONIA (NH_3) CONCENTRATIONS, TEMPERATURES, SALINITIES AND pH's

	TEST REGIME		
Concentration NH_3 (ppm)	18°C, 5 o/oo pH 8.0	20°C, filtered river, pH 6.5	21°, 5 o/oo pH 8.0
0	95	50	40
0.1	100	44	100
0.32	100	20	100
0.56	80	not tested	40
1.0	25	22	50
3.2	85	10	60
5.6	80	not tested	10
10.0	40	20	0

While the ammonia concentration for culture systems should not be in excess of 1 ppm for long periods (24 hours +), the use of some salinity (3-5 o/oo) would appear to enhance survival, especially at 18°C. Nitrate (sodium nitrate stock) concentrations of 0, 500, and 1000 ppm had little or no effect upon prolarval survival (70-80%) for nine days tested at 5 o/oo at both 18 and 21°C. However, the survival was reduced to 50 to 60% at concentrations of 1500 and 2000 ppm. Although none of the concentrations tested affected survival directly, only the controls (0 ppm) were observed to feed during the test period. To determine at what level below 500 ppm NO_3 this sublethal depression of feeding was active, groups of 60 just hatched prolarvae were exposed to concentrations 0, 56, 100, 560, and 1000 ppm NO_3 for 96 hours at 18°C. At the end of 96 hours exposure, they were removed to NO_3 free water at 18°C and fed. The mortality at the end of four days exposure was 20, 36, 22, 10 and 32% for the concentrations of 0, 56, 100, 560, and 1000 ppm, respectively. Six days after receiving their first food, all postlarvae at 1000 ppm were moribund and a few at 560 ppm were feeding. In the 100 ppm concentration many of the 70% surviving were feeding, while most of the 60% surviving at the 56 and 0 ppm concentrations were feeding and growing well. The survival in the 0 ppm concentration was complicated by fungal growth. Striped bass reared in a recirculating system have been reported to survive and grow through metamorphosis at nitrate levels of 34 to 141 mg/l (McIlwain, 1975).

Sazaki _et al_. (1972) reported results of impingement tolerance studies on striped bass larvae using chambers with screens of 16 or 30 meshes per inch. Larvae 10-15 mm were impinged at velocities in excess of 0.6 fps, and less than 20% were able to swim four minutes at 0.5 fps. These authors found that the 90% success level for swimming was 0.2 fps for larvae 10-15 mm and at 0.3 fps for larvae 20-30 mm. O'Connor and Schaffer (1977), using ichthyoplankton nets in an experimental flume at velocities of 0.5, 1.5 and 3.0 fps, observed that yolk sac larvae were most sensitive to velocities during netting followed, in decreasing order of sensitivity, by post yolk sac larvae and eggs.

Striped bass larvae are able to survive shear levels (laminar flow) of 350 dynes/cm^2 for one minute (Morgan _et al_., 1976). Mechanical damage to two-week-old larvae due to a single passage through a laboratory mock-up of a power plant condenser tube (excluding pump) was minimal (Coutant and Kedl, 1975). When temperature stress was added in this study, mortalities were comparable to thermal bioassay results. Different combinations of turbulent shear, pressure change and temperature rise were employed in this experiment. Finlayson and Stevens (1977) also observed that mechanical stress appeared to be the major factor causing mortality of entrained bass (8-31 mm). Their estimated TL_m was 31°C.

Biotic Factors

As Table 15 indicates, this is not an applicable factor for prolarvae which utilize the energy available in their yolk to develop their prey capturing ability. Larvae begin feeding during the later part of the yolk absorption period (Figure 11 c-d) and are known to be cannibalistic depending on availability of foods. Striped bass have been found to be relatively tolerant to food deprivation following yolk absorption (Figure 19). Unfed groups survived up to 22 days after hatching at 24°C and up to 32 days at 15°C. A "point of no return" does not appear to exist for this species (Eldridge _et al_., 1977; Rogers and Westin, 1979). Larvae age 9 to 14 days (after fertilization) exposed to heat shocks of 9-14°C above ambient (17-18°C) were found less likely to feed than control larvae not exposed to the heated water for 10 minutes (Van Winkle _et al_., 1979a). However,

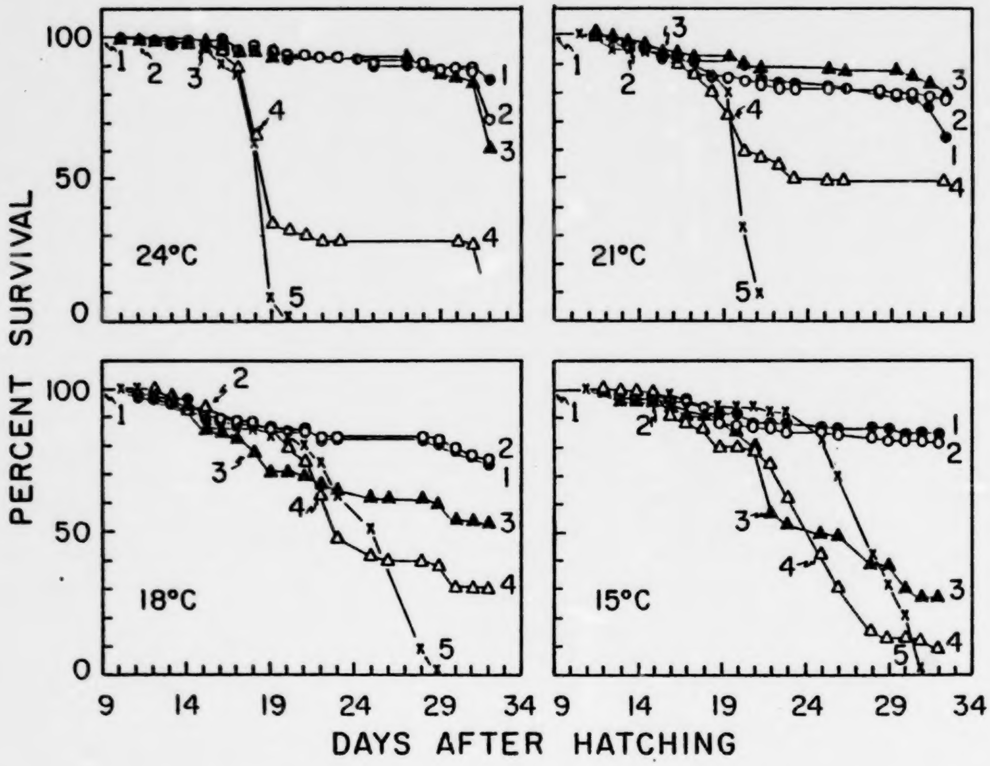


Figure 19. The effect of delayed feeding on the survival of striped bass stocked at yolk absorption at 24, 21, 18, and 15°C. Initial population was 100 larvae each. Numbered arrows indicate the order and time of first feeding. Group 5 was unfed throughout. (Rogers and Westin, 1980)

once heat shocked larvae recovered and began feeding, the amount consumed did not appear to be influenced by the exposures to heated water.

A food preference for pelagic species often over a more abundant benthic fauna, has been reported from field studies (Bowker *et al.*, 1969; Gomez, 1970; Humphries, 1971). Doroshev (1970) found that at mixed species food concentrations of 1000-1500 organisms per liter young bass (9-18 mm) had up to 35 organisms in their gut. Daniel (1976) estimated that to achieve the growth rate (in length) of early larvae observed in the estuary, his laboratory larvae would need to be exposed to food concentrations of almost 63,000 *Artemia* nauplii per m^3 daily for ten days. A comparison of survival and growth of prolarvae to 25 days fed on two diets was undertaken in hatchery troughs and aquaria. The report (Catchings, 1973) indicated that survival and growth were better (10.9% and 9.8 mm) for the brine shrimp diet than for the brine shrimp-dry feed diet (7.0% and 8.8 mm) during the period of the study. Experiments in a specially designed recirculating system of four diets formulated primarily of beef liver, or shrimp meal, two prepared diets (one Purina Trout Chow) and live brine shrimp nauplii were fed to larvae stocked as 4 day olds at 20°C for 10 days (Carreon, 1978). He found survivals of 95% for larvae fed brine shrimp and 76 - 45% (means of two replicates per diet) for larvae fed the other diets. Growth of brine shrimp fed larvae averaged 0.09 mm length daily, while growth on the other diets ranged from an average daily length of 0.02 mm to shrinkage of 0.05 mm. Braid (1977) also observed significantly better survival and growth of larvae fed live brine shrimp than those fed Chinook Trout Starter 1, Purina Trout Chow, Tetramin fish feed, freeze dried brine shrimp, or a microencapsulated feed. McIlwain (1975) reported starting four day old larvae on brine shrimp nauplii (about 20 per larvae twice per day) and introducing dry food (two parts commercial trout chow to one part pasteurized whole fish) into the diet when they were 11 days old. From this point the number of nauplii fed per day was decreased and the amount of dry food increased until they were feeding entirely on the dry food at 18 days old. Although 20-30 organisms twice a day are probably sufficient for first feeding larvae, we have determined that larvae will soon be consuming 10 times this amount. Individual larvae (each about 12 mm SL, 25 mg wet weight, and 2.8 mg dry weight) were fed brine shrimp (*Artemia*) nauplii twice daily for six days. Those at 18°C consumed 193 to 227 per day and grew from 1 to 3 mm SL during the six day period. At 21°C, the larvae consumed from 215 to 252 nauplii per day and grew 0.3 to 1.8 mm SL in the six days, while at 24°C, they consumed 292 to 305 per day and grew 0.3 to 0.8 mm SL. If an individual brine shrimp nauplii weighs 0.0016 mg dry weight (Paffenhofer, 1967), then 200 and 300 nauplii represents 0.32 and 0.48 mg dry weight, which is 12 to 17% of the average body weight of these larvae. We measured the *ad libitum* consumption of brine shrimp nauplii for 7-8 mm SL larvae at 22°C by weighing full and empty individuals. In this case, the larvae were found to consume up to 25% of their body weight (dry basis) at each feeding (twice daily). For a recently hatched 0.25 mg larva (dry weight) that would mean consuming 0.06 mg of food (dry) or about 40 *Artemia* nauplii (Paffenhofer, 1967) or 60 *Acartia* nauplii (Durbin and Durbin, 1978). For a larva of 2.3 mg dry weight this would mean consuming 350 nauplii. Miller (1978) estimated larval food consumption from preserved field collected bass < 10 mm TL with mean stomach volume of 0.37 mm^3, which he equated to about 154 copepod nauplii. His laboratory study showed that larvae (<10 mm) allowed to feed at a density of five zooplankters/ml contained an average volume of 0.26 mm^3 after 60 minutes at 21° C temperatures.

The effect of prey concentrations at one temperature have been reported by Al Ahmed (1978), Eldridge et al. (1977), and Miller (1977). In general, the survival response of larvae fed high and low food concentrations is similar to those fed early and late (Figure 19). For example, Eldridge et al. (1977) working at 18°C, reported survivals of larvae at 30 days after fertilization fed from day four at concentrations of 6.2, 4.0, and 2.2 nauplii (Artemia)/ml. The survival of their groups fed at 6.2 and 2.2 nauplii/ml corresponds at the same age to that of our groups (Figure 19) at 18°C first fed on day 19 (#3) to day 23 (#4) after hatching, respectively. The highest density Miller (1977) used was 3.6 nauplii/ml, but his survivals were only slightly better than that of starved controls (#5, Figure 19) of the same age at 18 and 21°C. That the survivals of all larvae fed earlier than day 19 (#1&2, Figure 19) at 18°C were 20% better than those of Eldridge et al. (1977) fed 6.2 nauplii/ml indicates that food densities of twice this may in fact be more realistic to ensure both maintenance and growth. The prompt onset of feeding following yolk absorption provides not only a survival advantage (Figure 19), but also a growth advantage which is not recovered by larvae reared at the same temperature but given their first food a few days later (Figures 20 and 21). Further, this rate of growth in weight and length following early and delayed feeding increases at a rate which is temperature dependent.

As the larvae grow, they require greater volumes of water in which to move and to ensure proper water quality. These constraints are applicable under culture conditions but not in their natural habitat. Densities of about 1000 prolarvae per gallon (or 250 per liter) have been suggested for holding in hatchery troughs during the first two weeks after hatching (Bonn et al., 1976). The densities indicated in Table 15 are approximations to the general 1 gm/l rule under semi-static conditions. The densities are shown in this Table as high to low concentrations to indicate that larvae should be graded and spread out as they grow. Since ammonia excretion and oxygen consumption are important factors in determining culture densities, they are discussed briefly.

Ammonia excretion was determined during this study on groups of prolarvae at 15 and 24°C using groups of 50 larvae per 300 ml with blanks for each. Ammonia was measured over the period of yolk utilization. The average excretion for six groups at 24°C was 0.636 ug $N\text{-}NH_3$ per larva per day. The average excretion for nine groups at 15°C was 0.245 ug $N\text{-}NH_3$ per larva per day through the prolarval period.

The oxygen consumption of a 9.4 mg bass in freshwater at 19°C was reported to be 2.05 mg O_2/gm/hr or 0.0135 mg/hr (Bodganov et al., 1967) We determined oxygen consumption of groups of prolarvae at 18 and 21°C at 5 o/oo. The larvae were 5-6 mm SL, and about 0.21 mg dry weight. The groups were of 20 larvae each in 300 ml of water with blanks. The mean

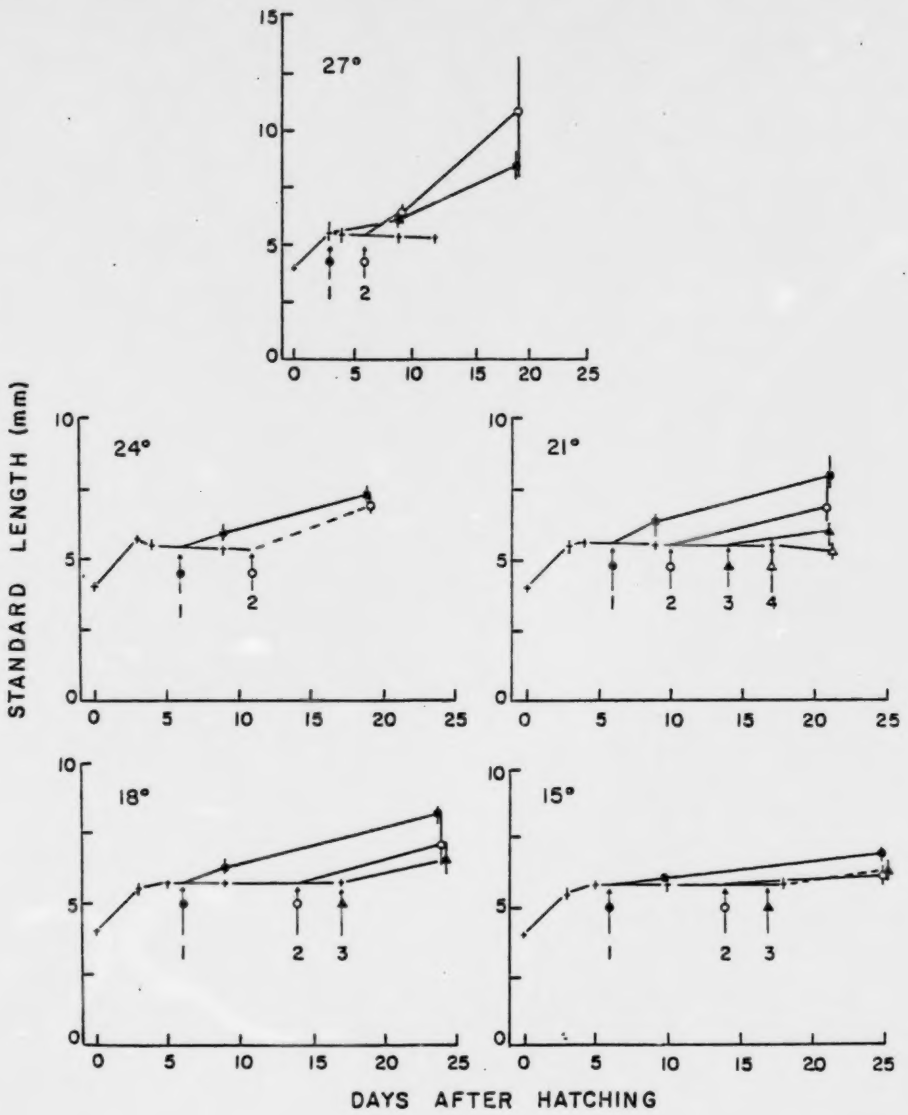


Figure 20. The effect of temperature and delayed feeding on the growth in standard length of striped bass larvae stocked at hatching at 27, 24, 21, 18, and 15°C. Each sample contains 10 individuals. Numbered arrows indicate time of first feeding for each population. Symbols identify groups which received their first food at the same time. The location of symbols denote sample means. Vertical bars indicate range of lengths in each sample. (adapted from Rogers, 1978)

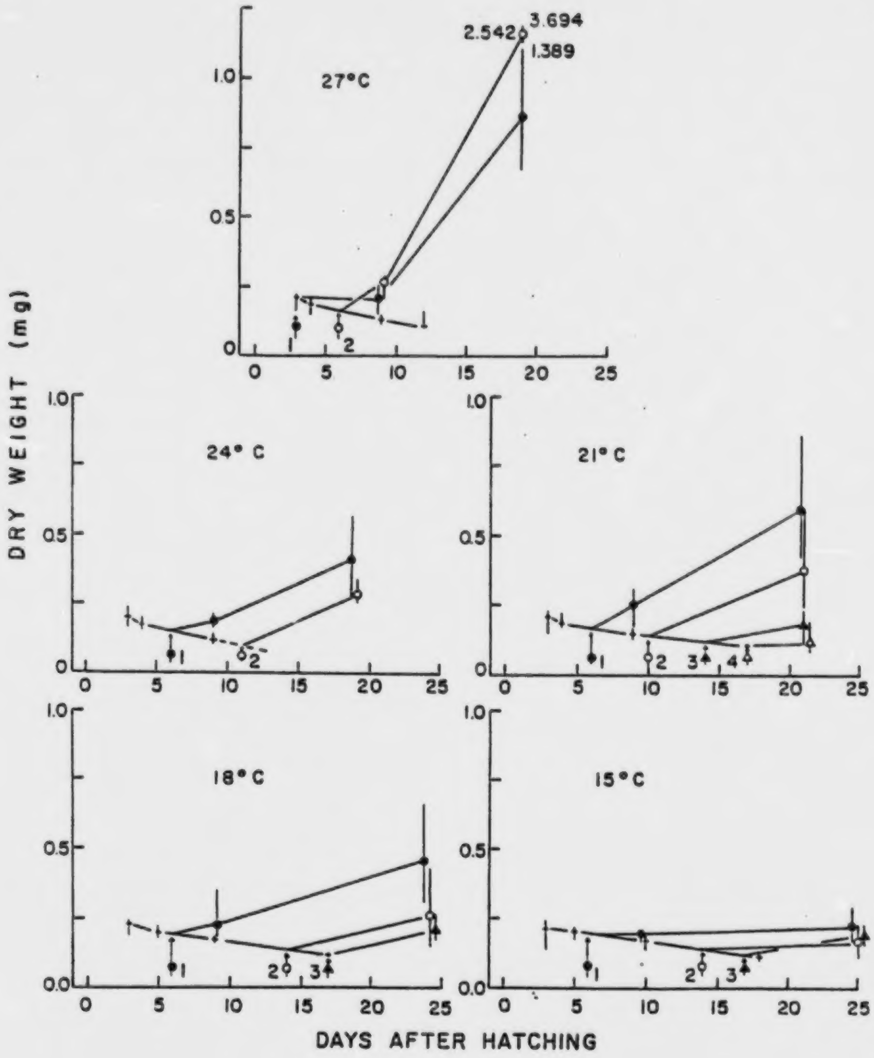


Figure 21. The effect of temperature and delayed feeding on the growth in dry weight of striped bass larvae stocked at hatching at 27, 24, 21, 18, and 15°C. Each sample contains 10 individuals. Numbered arrows indicate time of first feeding for each population. Symbols identify groups which received their first food at the same time. The location of symbols denote sample means. Vertical bars indicate range of weights in each sample. (Rogers and Westin, 1979)

oxygen consumption for four groups at 18°C was 0.86 ul/hr/larva. These rates were for larvae utilizing yolk and not feeding. A single group measurement of 20 larvae (7-8 mm and 0.36 mg/dry weight) each fed prior to the measuerments showed rates of 1.50 ul/hr/larva at 18°C and 3.10 ul/hr/larva at 21°C.

In their natural habitat striped bass larvae have many predators - including older striped bass. In culture systems cannabilism has also been observed. We have noted that its incidence is much reduced when excess foods of varied sizes are present and when the size hierarchy of the rearing container does not become extreme.

Diseases and parasites include any of those reported for juveniles and are discussed in the next section. Fungus can be a problem as with eggs, although increasing the salinity to 5 o/oo will reduce it. Salinity of 10 o/oo reduced predation by microhydras on larval bass in a laboratory situation (Dendy, 1979). Larvae can also be treated with malachite green at 0.05 to 0.1 mg/l as a dip, if necessary.

CULTURE METHODOLOGY

Capture Methods

The most successful capture method is the hatching of eggs already under culture. Production for pond stocking, especially popular in the southeastern United States (Braschler, 1975; Bonn *et al*., 1976), is carried out by a number of state and federal hatcheries. This was discussed in Section 4.

Hatching is the recommended capture procedure since it does the least damage to the survival of the larva, which has neither the protection of a chorion nor scales.

Post-Capture Handling

In the hatcheries (state and federal) the larvae are allowed to swim up (yolk and oil giving bouyancy) in the hatching jars with the freshwater flow and spill over into 30 gallon aquaria. These aquaria have a standpipe drain surrounded by a perforated metal screen. Up to 1,500,000 prolarvae can be held in these aquaria provided the rate of water exchange is one gallon per minute (Bonn *et al*., 1976).

If the larvae are recently arrived (i.e., transported from the place of hatching to another culture facility), a close look before stocking into the receiving water is warranted. Before stocking is the time to treat, if necessary, for fungal or bacterial infections. A dip with malachite green (0.05 to 0.1 mg/l) is recommended and can be accomplished while the shipping container water and larvae come to receiving water temperature. When receiving transported larvae, it is good practice to aerate their shipping water while waiting for the temperatures to equilibrate.

Transportation

Larval shipping procedures, described by Bayless (1972), Bonn et al., (1976) and Texas Instruments (1977c) are essentially as follows. The larvae, concentrated by removing water from the holding container, are dipped into a plastic bag fitted into a styrofoam box. Water is added to the bag until it has half filled the volume of the shipping box. Oxygen is added to the shipping water and allowed to fill the space over the water. The bag is sealed with double castration bands and the box top taped in place. The general density for shipping is estimated at 40-50,000 per gallon, or about 13,000 per liter. Although most of the hatcheries use their well-water (fresh) source for shipping, we have found that additions of filtered sea-water to bring the salinity to 5 o/oo, or the addition of penicillin-streptomycin as described in the embryo section improve water quality and survival during the shipping period. Larvae should not spend more than two days in shipment, especially if the temperature rises above about 18°C. Upon receipt of the larvae, the box and bag should be opened, the temperature recorded and the larvae inspected by dipping some out. They should receive aeration while observations of their condition are made and the shipping-receiving waters equilibrate.

Handling Procedures

Care should be taken when handling larvae not to remove them from the water. The tools shown in Figure 9A for handling eggs should be used for larvae. Nets should not be used until the larvae are fully scaled (metamorphosis). Larvae can be weighed and measured, but this is usually on a sacrificial basis, at least until anesthetics can be used. Larvae will respond to anesthetic variably until after yolk absorption and successful feeding (Figure 11 e-g). After this they can be anesthetized with certainty that they will recover, if they are not abused during the measurement period. The anesthetic we found most successful was MS-222 at 50 mg/l concentrations.

In each culture system some larvae will grow slightly faster then others. We noticed that the larger, faster growing larvae remain nearer the bottom, while the smaller, slower growing larvae, are more often nearer the surface. It is generally recommended that the larvae in the system be graded to keep the size hierarchy within any one tank to a minimum. This will improve the survival of the smaller larvae by reducing the chances for cannibalism. The grading system we found to be most conducive to larval survival was also the most time consuming. It consisted of lowering the water level in the tank until all fish were visible and with glass beakers dipping the selected fish into a holding container. At times it was easier to remove only the larger ones and leave the smaller ones in the tank which was then refilled with culture water. At other times, the quickness of the larger fish made it easier to remove all of the smaller fish to another tank. This is also a very good time to observe the growth and condition of the larvae in the culture tanks easily.

Maintenance Procedures

Culture Vessels--

A variety of culture vessels has been used in large scale larval rearing. Most of the hatcheries in the southeastern United States rely on flowing freshwater for their systems which may involve rectangular tanks, aquaria or screen cages in the ponds to rear the larvae prior to stocking in ponds at 6 to 20 days after hatching (Bonn et al., 1976). Several recirculating or closed filter system and tank combinations have been utilized. Rhodes and Merriner (1973) described a culture system composed of a 10 foot in diameter pool (900 gallons) with three filter pans of fiberglass wool and activated charcoal. Their system was used to rear prolarvae successfully through metamorphosis at 0-6 o/oo and 17-27°C. McIlwain (1975) reported using a series of 1000 gallon rearing tanks connected to a filter tank of shell material (Figure 22A) in which prolarvae were successfully reared through metamorphosis at 2 o/oo. Lewis and Heidinger (1976) favored an upflow rearing tank (Figure 22 B) which was part of a large scale closed culture system equipped with both rapid sand filters and biofilters on a well-water source.

The system we utilized for most of our larval rearing was similar to the semi-recirculating system described by Houde and Ramsey (1971). This system had not been used for striped bass larvae, although it had been used with success for many marine fish species. We found this static prepared tank rearing system successful. It has the following advantages which we feel make it particularly well suited for laboratory rearing: 1) it is uncomplicated; little or no complex equipment is required; 2) it is relatively immune to weather and temperature changes which plague systems relying on natural water supplies; 3) the life support system is an integral part of the culture water, and fish do not have to be separated using screens so that the water can be treated and returned; 4) dissolved oxygen may be maintained at a high level without excessive aeration. Disadvantages include: 1) the system may be unsuitable for high volume hatchery operations; 2) some care must be exercised to prevent introduction of foreign algal species.

Culture containers consisted of 55 gallon (208 l) polyethylene drums and 175 gallon (662 l) fiberglass tanks. Over each container at a height of about one foot were mounted banks of fluorescent lamps. The light levels used, four forty-watt cool white bulbs per 175 gallon container and the equivalent of two per 55 gallon container, could probably be halved. Light was supplied continuously. Culture water was made by mixing tap water delivered through non-metalic plumbing with filtered sea water to a salinity of 6 to 8 o/oo. This salinity was used with the following considerations in mind: a) it was well within the tolerance range of striped bass larvae; b) it prolonged the life of the brine shrimp nauplii supplied as food; c) it reduced the probability of *Saprolegnia* proliferation; and, d) it was a more amenable salinity for the flagellate species used to maintain the system.

The algal species used in the system was a euryhaline, eurythermal tide pool flagellate, *Brachiomonas*, which was supplied by Dr. Paul

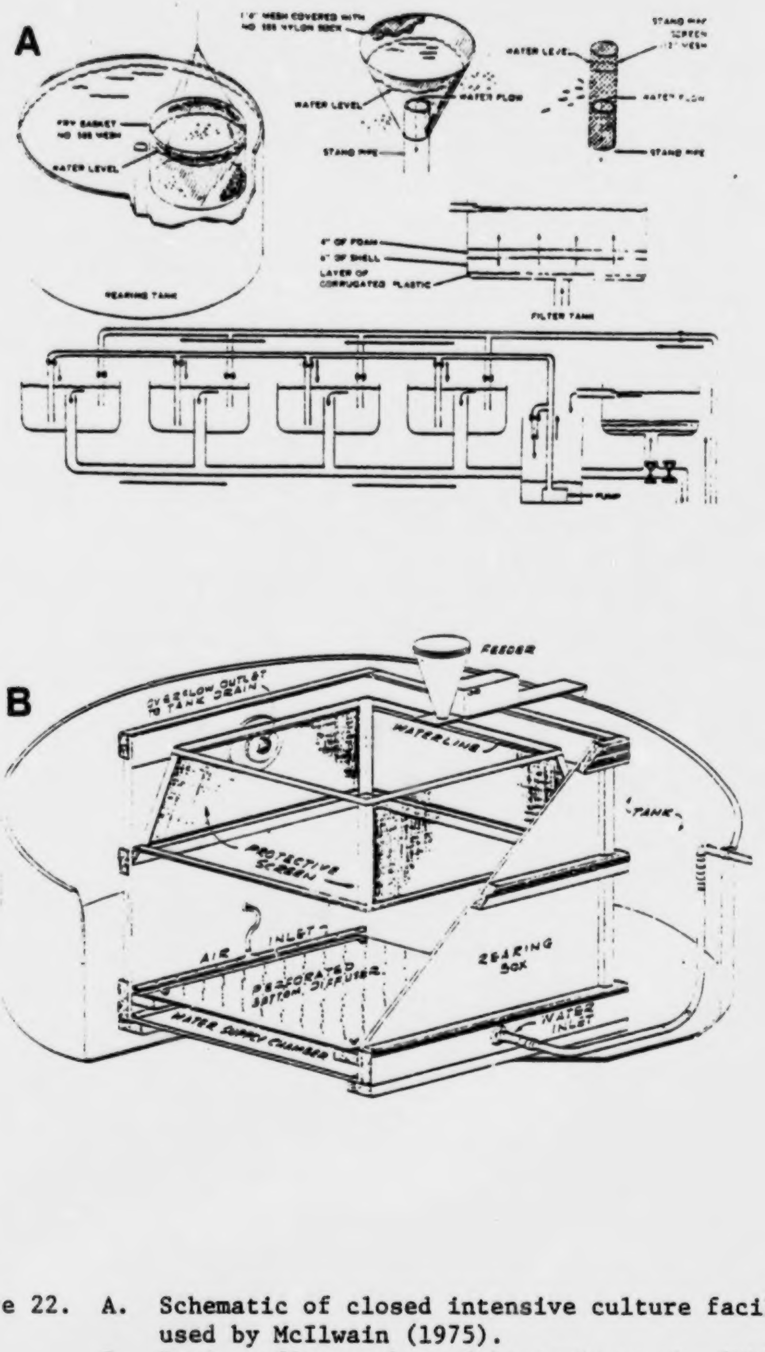

A
1'0" MESH COVERED WITH
NO 355 NYLON SOCK
WATER LEVEL
WATER FLOW
STAND PIPE
STAND PIPE
SCREEN
1/2" MESH
WATER LEVEL
WATER FLOW
STAND PIPE
FRY BASKET
NO 355 MESH
WATER LEVEL
REARING TANK
4" OF FOAM
6" OF SHELL
LAYER OF
CORRUGATED PLASTIC
FILTER TANK
PUMP
B
FEEDER
OVERFLOW OUTLET
TO TANK DRAIN
WATERLINE
TANK
PROTECTIVE
SCREEN
AIR
INLET
PERFORATED
BOTTOM DIFFUSER
REARING
BOX
WATER SUPPLY CHAMBER
WATER
INLET

Figure 22. A. Schematic of closed intensive culture facilities used by McIlwain (1975).
B. Basic upflow tank used by Lewis et al. (1977) for bass larvae in their recirculating system.

Hargraves of the University of Rhode Island. In its natural habitat this species is subjected to rapid changes in temperature and salinity. Pools are frequently enriched with bird droppings, hence this species is tolerant of and capable of using ammonia, a prime metabolite of both larval fish and brine shrimp, as a nitrogen source. Flagellates were chosen because, being motile, they would remain throughout the water column and not sediment to the bottom of the tank. Their rapid generation time allowed them to respond to changes in nutrient level in the system.

Low level aeration was supplied to keep the culture well mixed. The culture water in each tank was enriched to the proportions of Guillard's F/2 algal culture medium (Guillard, 1975). These nutrient levels were non-toxic to the larvae and promoted algal growth. Deleted from the algal medium was any nitrogen source. This was supplied by the larvae and their food. As the larvae developed and were graded and the algal medium in the system was renewed, the salinity was increased so that metamorphosing larvae were at 25 o/oo. As the larvae reached metamorphosis, they were transferred into running sea water systems.

Stocking Density--

The stocking density reported by McIlwain (1975) was 5-60 prolarvae per liter in the recirculating system. Rhodes and Merriner (1973) suggested that stocking in a system such as the one they described should be limited to 100,000 per 900 gallons, or about 30 per liter. Lewis and Heidinger (1976) reported stocking densities of 55 to 182 prolarvae per liter in the various tanks in their recirculating system. Rogers et al. (1977) stocked 30 to 150 prolarvae per liter in their static experimental containers. Bonn et al. (1976) suggest an optimum stocking rate for ponds at 100,000 per acre for post yolk sac larvae and recommend that prolarvae be held at a density of 100,000 per cubic meter (100 per liter). Based on information presented above, 100 early post larvae stocked into a liter at 18°C can be expected to excrete at least 0.4 mg N-NH_3 into and consume about 3.6 ml of oxygen from that liter daily. This is equivalent to 5.1 mg/l or about half of the saturated level of dissolved oxygen. Hence, stocking in excess of 100 larvae per liter is not recommended after feeding initiates. Stocking rates should probably be 50 larvae per liter at most, especially as the larvae develop.

Maintaining Water Quality--

In recirculating systems water quality is often poor because of filter inefficiency and the accumulation of organic matter in the water due partially to improper sumping (Lewis et. al., 1977). Increased organic particulates (food and feces) may decrease the dissolved oxygen concentration by increasing the total, biological oxygen demand of the rearing water. In our algal system, we periodically replaced a portion of the algal population which was approaching senescence, thereby returning the culture to more active log growth. Any algal contaminates, such as diatoms, were removed before they could decay and cause deterioration of the water quality. The continued light was found to be unnecessary to ensure maintenance of near saturated dissolved oxygen levels. Short dark periods (approximately 4 to 6 hours) were found useful in maintaining the algal system pH at 7.5.to 8.5 range. The recommended water quality for larval rearing should include dissolved oxygen levels at or near saturation, ammonia levels <0.5 to 1.0 ppm, nitrate levels (especially in recirculating systems) of <56 ppm and a pH of 7-8. We recommend raising salinity and temperature during the larval stage from about 5 o/oo and 18°C for prolarvae to 5-10 o/oo and 20°C for post yolk-sac feeding larvae to 20-25 o/oo and 22-25°C near metamorphosis. Under this regime metamorphosis should be attained within two months of hatching.

Diet--

Although most of the work has been done using just brine shrimp (Artemia) as the exclusive diet, there are several reports of feeding larvae on brine shrimp with some percentage of dry food added. Rhodes and Merriner (1973) fed their larvae brine shrimp (90-50%) plus up to 50% Tetramin flakes, which were observed to foul the rearing water. McIlwain (1975) reported success in weaning larvae to an all prepared dry diet of commercial trout chow and pasteurized fish (approximately 50% protein and 7.2% fat). This diet did not apparently effect the filter capacity and/or water quality and was moderately acceptable to the larvae. However, a number of studies mentioned earlier in this section showed that better survival and growth of larvae resulted on diets of live brine shrimp rather than on prepared diets. In our systems, larvae were exposed to Artemia nauplii well before they were capable of feeding to be certain food was available when it was needed. For the first several weeks after hatching nauplii were supplied in excess to each container twice a day. Uneaten nauplii were always present at the next feeding. Later as the cumulative appetite of all the larvae in each container increased, larger brine shrimp were fed and feedings were more frequent. Although some bass failed to feed and died within 10 days of their arrival, most commenced feeding without difficulty. Their bodies could be seen to become pink with nauplii shortly after each feeding. We also used wild zooplankton species as supplements. Table 17 presents the caloric and percent composition of some of the live diets used for larvae.

Digestion time influences the amount of food a larva can consume. Al-Ahmad (1978) observed that at 25°C larvae (8, 13, and 18 days old) had digested the rotifers they consumed during a one hour period in 3 to 6 hours. Larval egestion was reported as 11 to 12 hours at 20°C for 15 and 19 day old larvae and less than 9 hours at 25°C for 9 day old larvae fed brine shrimp (McHugh and Heidinger, 1977). Eldridge et. al.(1980) reported that digestion of brine shrimp varied with the size of the larvae from 1 to 5 hours at 18°C. We observed groups (10-15) of larvae (3-5) and individuals (10-20) at each of three constant temperatures to estimate digestion times. The brine shrimp fed provided a natural marker (pink in the gut) and the transparency of the larvae allowed us to make observations on full larvae at intervals of 20-30 minutes until they were empty. Digestion times observed were 5-6 hours at 18°C, 4-5 hours at 21°C and 3 hours at 24°C.

Information presented earlier indicates that larvae can easily consume 10-20% of their dry body weight per meal. If they digest this meal in 4-5 hours, then feeding twice per day is probably just sufficient and any less might lead to starvation. Of course, feeding more often would promote both

TABLE 17. CALORIC AND PERCENT COMPOSITION OF SOME LIVE LARVAL FOOD ITEMS

Food Item	Calories/gram (ash-free, dry)	Percent of Dry Weight lipid	Percent of Dry Weight protein
Artemia salina			
nauplii	5800-6000(1)* 5454-5953(3)	15.04-27.24	42.5-50.2(1)
adults	5115-5854(3)	6.51	62.78(1)
Acartia clausi		5.8	82.6(4)
Acartia tonsa	5664 ± 86(2)		
Calanus finmarchicus	6835 ± 191(2)	10.5-47.0	30-77(4)
Calanus helgolandicus	5515 ± 277(5)	11.0	75.2(4)

* Number in parenthesis refers to source: (1) Helfrich *et al*. (1973); (2) Laurence (1977); (3) Paffenhofer (1967); (4) Raymont *et al*. (1963); and (5) Slobodkin and Richman (1961).

survival and growth, provided water quality was maintained. Consuming 200 nauplii each, 50 larvae stocked per liter would require 10,000 nauplii per liter. This is almost twice the highest concentration (6.2 nauplii/ml) tested to date (Eldridge et al., 1977). As the larvae grow, it is reasonable to increase the size of their live diet until they can feed on ground fish, squid, or prepared diets after metamorphosis. The diet chosen for larvae should not contain less than about 43% protein and 5700 cal/gm dry weight. These are values comparable to the caloric value of yolk (see Section 8) which the prolarva utilizes initially.

Normal Conditions and Physiological State

Larval growth at five temperatures through the first 20 days after hatching is shown in Figures 20 and 21 for length and weight. Instantaneous growth coefficients on a dry weight basis for the larval groups plotted in Figure 21 are given in Table 18 . The growth rate through metamorphosis on the basis of larval length is shown in Figure 23 . This figure combines growth curves from several studies at a variety of temperatures under "excess" rations. Compared to the growth rate attributed to "wild" bass (temperature unspecified) of Humphries and Cumming (1973), the other three laboratory studies (Mansueti, 1958 at 15 to 18°C; Rhodes and Merriner, 1973 at 17 to 27°C; Rogers et al., 1977 at 15 and 24°C) appear to underestimate the growth rate of striped bass in nature.

Prolarvae normally drift in a head-up position, because of the bouyancy and location of the yolk sac (Figure 11a). They make short erratic swimming movements at this stage. Movement becomes more vigorous as the larvae absorb their yolk material. Larvae tend, therefore, to move passively with any currents during their prolarval period. Post yolk sac larvae (Figure 11 d-e), however, are strong swimmers by comparison. Activity patterns among 10 and 25 mm larvae exposed to water velocities from static to 27 cm/sec were observed in the presence and absence of food (Bowles, 1976). Visual cueing apparently played an important part in feeding among these larvae. The dominant orientation for 25 to 80% of all of the observations in this study was swimming into the current (positive reotaxis).

Normal development proceeds as indicated in Figure 11 and Table 13 for larvae. Larval condition may be checked by sampling and comparison of the larval length and dry weight to that given by the equation with the description of this stage. Histological and morphological changes during starvation have been described for several larval fish species (Ehrlich et al., 1976; O'Connell, 1976; Theilacker, 1978). The criteria developed to assess the nutritional condition of these species can probably be used to generalize starvation conditions of larval striped bass. The digestive tract is apparently the first area to show tissue atrophy. During starvation in the species investigated growth was retarded, the larvae shrank and the soft tissues collapsed, causing the larvae to appear abnormal. Shrinkage of starved striped bass in both length and weight can be seen in Figures 20 and 21. A number of the abnormalities reported among fed bass larvae, some of which may in fact be starvation related, are shown in Figure 24 . One abnormality shown (A, bottom; B second from top) and

TABLE 18. INSTANTANEOUS GROWTH COEFFICIENTS FOR DELAYED FEEDING GROUPS AT FIVE CONSTANT TEMPERATURES

Temperature (°C)	Initial Dry Wt. (mg)*	Final Dry Wt. (mg)	Days Since Hatching: Day of First Feeding	Days Since Hatching: Day Measured	Instantaneous Growth Coefficient**
27	0.211	0.863	3	19	8.803
	0.157	2.542	6	19	21.419
24	0.155	0.413	6	19	7.538
	0.100	0.289	11	19	13.266
21	0.170	0.593	6	21	8.330
	0.140	0.376	10	21	8.981
	0.120	0.180	14	21	5.793
	0.102	0.111	17	21	2.114
18	0.190	0.451	6	24	4.802
	0.135	0.257	14	24	6.438
	0.116	0.205	17	24	8.135
15	0.198	0.231	6	25	0.811
	0.145	0.174	14	25	1.657
	0.125	0.195	17	25	5.558

*Determined by interpolation when the actual weight was not available (see Fig. 21)

**Instantaneous growth coefficient (Ricker, 1971).

$$\frac{\log_e wt_2 - \log_e wt_1}{t_2 - t_1}$$

where wt_1 and wt_2 are dry weight at times t_1 and t_2, respectively.

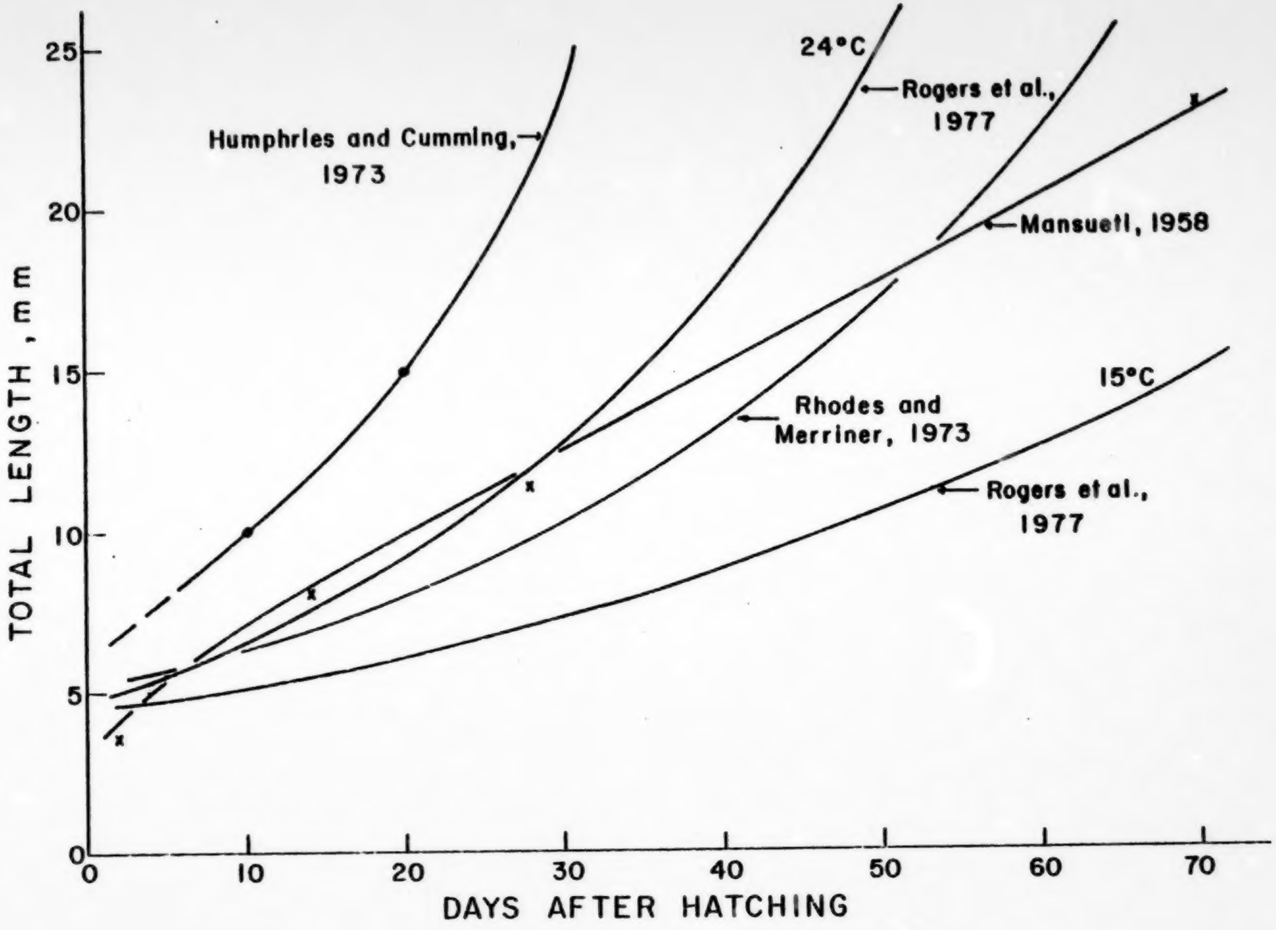


Figure 23. A comparison of growth rates observed under fixed temperature regimes (Rogers et al., 1977) with those obtained in earlier studies under conditions of increasing temperature.

reported frequently from hatchery situations is the non-inflation of the swim (or gas) bladder. Bulak (1976) reported that food availability and heat stress had no effect on the time of initial inflation. It appears that reduced oxygen in the water is the major factor in the failure of the swim bladder to inflate (Bulak, 1976; Doroshev and Cornacchia, 1979).

Diagnosis and treatment of pathological conditions occurring in larvae have been mentioned above or are discussed in the section describing juvenile diseases.

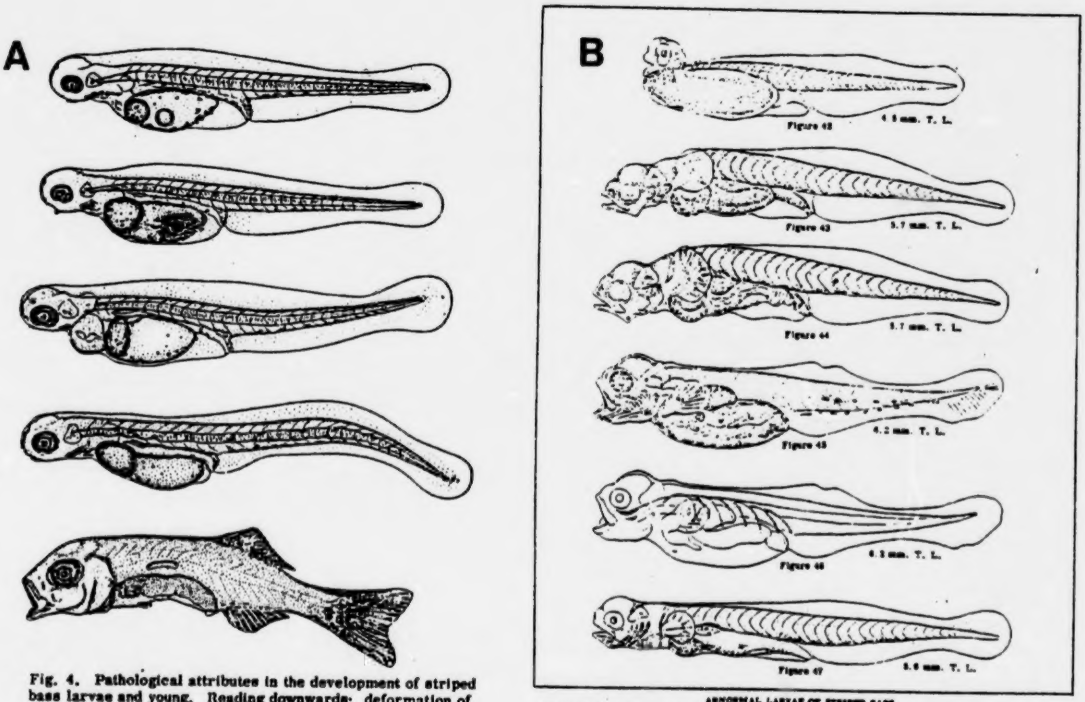


Fig. 4. Pathological attributes in the development of striped bass larvae and young. Reading downwards: deformation of the oil droplet in a 3-day-old larva; deformation of the yolk sac at the same age; inundated pericardial sinus, age 2 days; distortion of the notochord, age 4 days; gas bladder not filled with air, age 35 days.

ABNORMAL LARVAE OF STRIPED BASS

FIGURE 42. Abnormal prolarva, 4.5 mm. long, showing constricted head.
FIGURE 43. Abnormal postlarva, 5.7 mm. long, showing humpback deformity.
FIGURE 44. Abnormal postlarva, 5.7 mm. long, showing humpback and enlarged oil globule.
FIGURE 45. Abnormal postlarva, 6.2 mm. long, with blue-sac disease.
FIGURE 46. Abnormal prolarva, 6.2 mm. long, with blue-sac disease, showing blisters, blood clots and enlarged body grooves.
FIGURE 47. Abnormal postlarva, 5.6 mm. long, showing an almost total lack of pigment.

Figure 24. Abnormalities among striped bass larvae reported by: A - Doroshev (1970) and B - Mansueti (1958).

SECTION 10

RECOMMENDED CULTURE METHODS AND BIONOMICS: JUVENILE AND SUBADULT

DESCRIPTION OF STAGE

This stage spans that portion in the life history of a striped bass from metamorphosis to maturity. It is the adolescent stage during which the young bass resemble the adult in scalation, form, coloration and general behavior. From metamorphosis to their first birthday (June 1 for ageing purposes), bass are referred to as juveniles, young-of-the-year (YOY), fingerlings, or age group 0. From the first birthday to the second, they are generally referred to as yearlings (age group I), and during the year from the second to the third birthday as two-year-olds (age group II).

Metamorphosis, the end of the larval stage, begins at about 15-18 mm total length (TL) for preserved specimens (Rogers et al., 1977). This corresponds well to a live equivalent standard length (SL) when shrinkage due to preservation is considered. The juvenile stage is considered to begin at 20-30 mm SL, 110 to 400 mg live weight and 20-75 mg dry weight. This subadult stage extends to maturity, which varies with sex. Many males mature during their second year and all are mature by their third year at a length of approximately 250 mm or more. The majority of the females mature during their fifth and sixth years, at a length of 500 mm or greater. Maturity is considered to be at a minimum of 300 mm, about 350-400 g live weight and about 90-160 g dry weight. Figures 25 and 26 provide a general idea of the expected normal ranges of lengths and weights for live juvenile and subadult striped bass.

Similar length-weight relationships (see Table 29) have been reported for juveniles from the Hudson River, Rappahannock River, and Albermarle Sound. Often it is not standard length (SL) that is available, but fork length (FL) or total length (TL). Trent (1962) found that for juveniles (20-100 mm TL) the relationships were: $FL = 0.93835TL - 0.077817$; $SL = 0.80388TL + 0.55750$; $SL = 0.84675FL + 1.22099$. From our measurements we found that $SL = 0.909FL - 1.805$ and $FL = 1.55SL - 0.196$. Mansueti (1961) used a factor of 0.93 to convert TL to FL for subadults and adults, while Texas Instruments (1973) found that $FL = 4.60 + 0.902TL$ for subadults.

During this study we determined the caloric content of adolescent striped bass to be 5350.8 cal/g ash-free dry weight (n=3; range 5146.4 - 5592.2) with a mean ash content of 17.3% (9.8 - 21.4) of the dry weight. The percent water ranged from 69 to 79% with a mean of 74.6% (n=102) for bass throughout the size range of Figure 26.

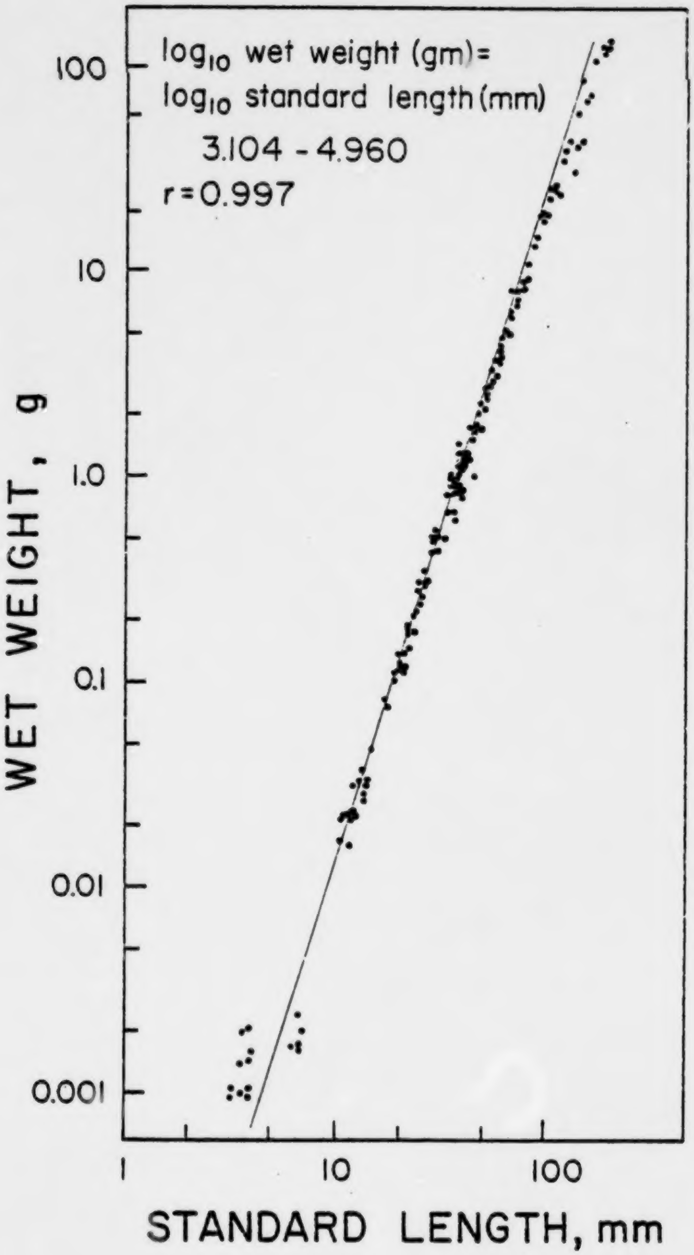


Figure 25. Relationship between standard length (SL) in millimeters and body weight in grams for post yolk sac larval and juvenile striped bass. The regression equation was calculated from transformed data.

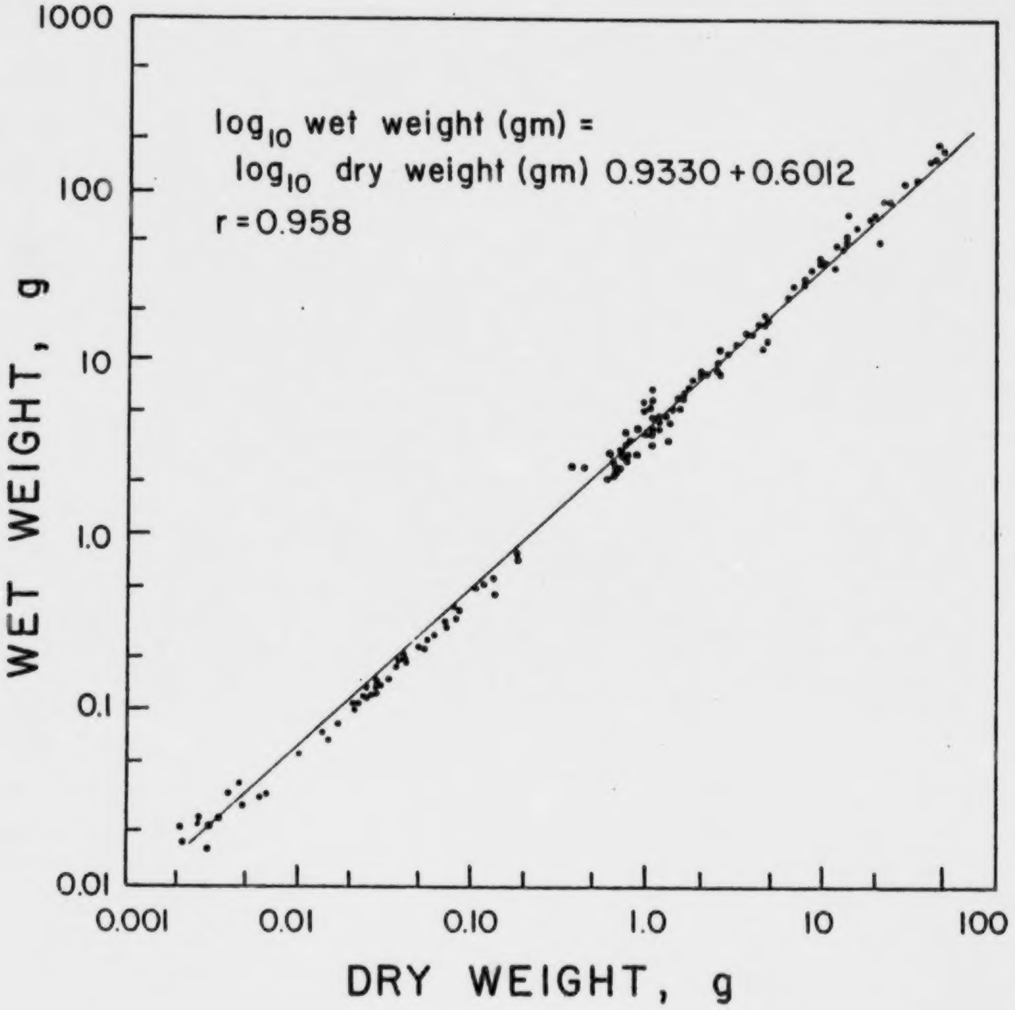


Figure 26. Relationship between dry weight in grams and wet weight in grams of juvenile and subadult striped bass. The regression equation was calculated from the transformed data.

Wood and Hintz (1971) investigated the stability of striped bass lipids during storage at ice temperatures. They noted the phospholipid fraction contained the highest proportion of polyunsaturated acids and the neutral lipid fraction contained the highest proportion of monosaturated acids in fresh tissue. The polyunsaturated acids $C_{20:5}$ and $C_{22:6}$ were most affected during storage and were lost at a faster rate from the phospholipids than from the neutral lipids. Phospholipids accounted for 40% of the total body lipid composition of feeding juveniles (Dergaleva and Shatunovskiy, 1977). Korn and Macedo (1973) determined that 2 g striped bass were 18.2% fat by Goldfisch and column fat extraction techniques. Iodine numbers (a measure of the relative heat stability of fat) for juvenile and yearling bass ranged from 123 to 189 (Loeber, 1951).

Blondin et al. (1966) showed that sterol biosynthesis in striped bass follows the same pathway as demonstrated by others for mammals, but occurs at a significantly slower rate in bass. They reported that the primary sterol found in bass liver is cholesterol. Squalene, lanosterol and cholesterol were identified as metabolites of mevalonic acid. Blondin et al. (1967) found the rate of vitamin D formation to be small compared to the rate of chlolesterol formation in striped bass liver.

NATURAL HABITAT

Juvenile striped bass are abundant in spawning areas and more saline nursery areas about two months after spawning occurs. Local movements of these juveniles and yearlings have been well documented in areas of proposed power plants (Hudson River, Chesapeake-Delaware Canal and Potomac River) or pump storage and canal diversions (Sacramento-San Joaquin River Valley) and were described earlier in Section 6. Juvenile bass are first collected in mid-June to early July, depending on time of spawning, from river waters deeper than 6 meters (Figures 27 and 28). As the water temperature increases, the juveniles migrate to shoal and shore zone areas. Falling water temperatures bring net downstream movement so that by December juveniles are generally absent from the shore zone, having either left the estuary or moved into deeper water for winter. Apparently, the abundance of juveniles in local areas is related to temperature, salinity, habitat type, diel patterns, and tidal stage.

Other factors have been postulated to influence juvenile abundance in addition to temperature. Three factors - mortality, dispersion, and gear selectivity - were presumed responsible (separately or in combination) for a reduction seen in young-of-the-year abundance as the season progressed (Trent, 1962). Observed migrations of young-of-the-year and juvenile bass downstream from the Sacramento-San Joaquin Delta (Sasaki, 1966) probably took place in response to food supply and/or water velocity changes. Survival and distribution of young bass were clearly defined functions of water flow in this delta system and abundance was greatest in the low salinity zone (Turner and Chadwick, 1972). Possible mechanisms for these relationships were discussed by the authors. A more detailed discussion of factors affecting abundance is presented in Section 13.

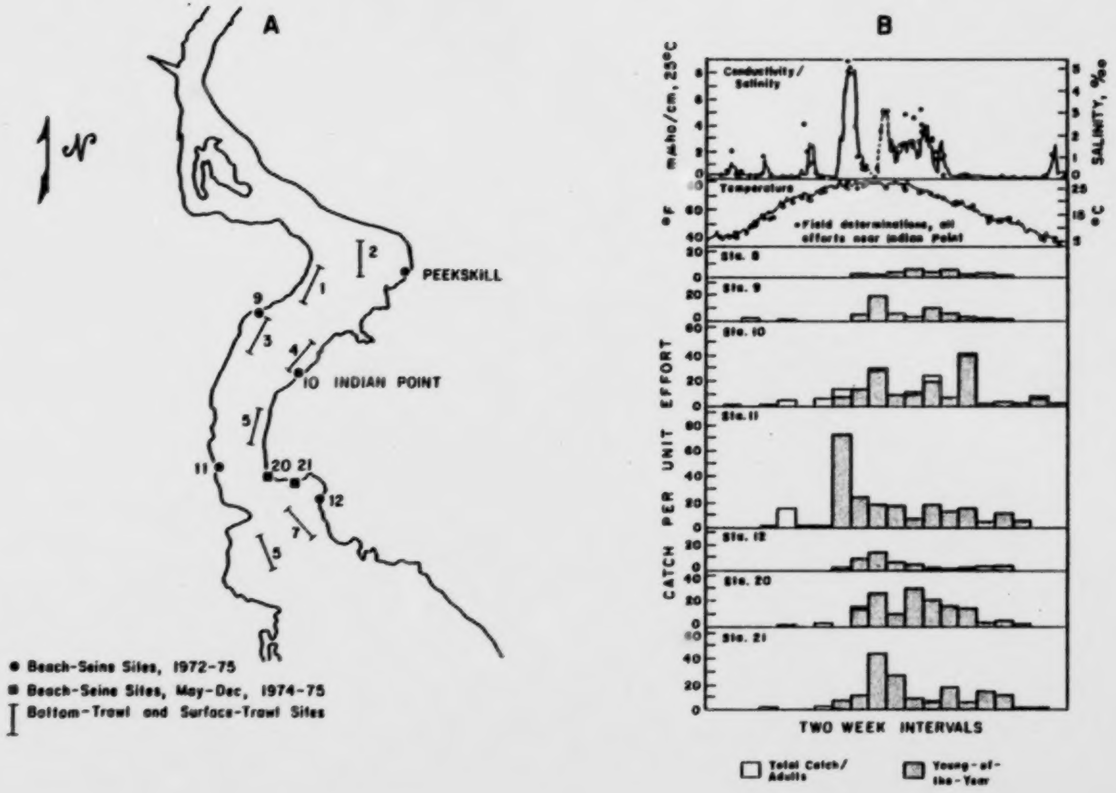


Figure 27. Hudson River sites (A) for juvenile striped bass catch per unit effort (B) at two week intervals beginning at 3/23-4/5 through 12/14-12/27. (adapted from McFadden, 1977a)

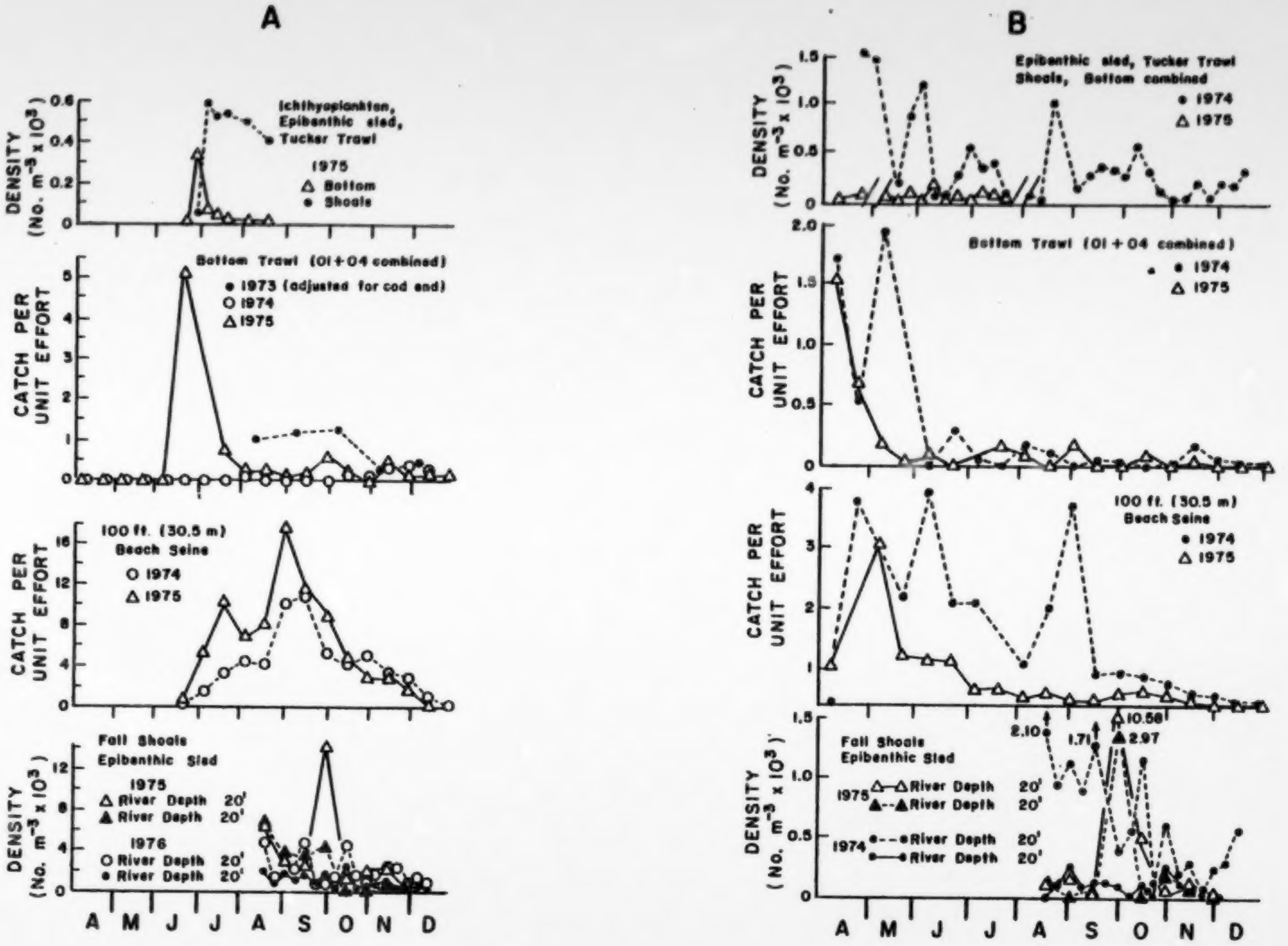


Figure 28. Abundances of striped bass juveniles (A) and yearlings (B) taken in standard samples during 1973, 1974 and 1975 from the Hudson River. (adapted from McFadden, 1977a)

Striped bass two or three years old are not generally involved in coastal migrations, but they do form schools, moving about their river or estuary area. The young-of-the-year bass also move in large schools within the river of their spawning. These schools appear to over-winter in deeper sections of the river. It is these schools of subadults (ages II and III) which contribute substantially to the commercial and sport catches (see Section 14) in the spawning rivers (Frisbie and Ritchie, 1963; Grant, 1974; Shearer et al., 1962; Tiller, 1950).

ENVIRONMENTAL REQUIREMENTS

Table 19 summarizes the environmental requirements of this stage. Background information on these factors is presented in more detail below.

Abiotic Factors

Juvenile bass were observed to tolerate 9°C rise above normal temperatures of 22°C (Kerr, 1953). The maximum upper temperature avoided by juveniles in the summer (27.2°C acclimation) was 33.9 and 34.4°C, while the maximum avoided from 5.0°C acclimation was 12.8°C (Meldrim and Gift, 1971). No consistent relationship between salinity or light level and upper avoidance temperature was observed by these authors. The minimum temperature at which we have observed survival was 0 to -1.0°C at our winter ambient sea temperatures. Fingerling bass (30-70 mm TL) survived tests at 32.2°C after reacclimation for 12 hours from 16 to 26.7°C, however all fish died when the temperature exceeded 35°C (Davies, 1973). Texas Instruments (1976b) found that the median thermal tolerance limit (TL_m) for YOY and yearling bass (39-230 mm TL) changed with changing temperature acclimation throughout the year. During the period of falling Hudson River temperatures (26 to 11.5°C) the TL_m declined from 34°C to 28.6°C, and during the period of rising river temperatures (15.5 to 26°C) the TL_m increased from 29 to 34°C. They also observed 100% mortality among yearling bass during 96 hours after a temperature drop from 15 to 2°C. However, none of the bass tested showed loss of equilibrium or death with a drop from 10 to 2°C. These juveniles failed to avoid lethal temperature conditions when acclimated to temperatures less than 9.5°C in Hudson River water. The upper avoidance temperature was 22.5, 29.0, and 32.0°C when the bass were acclimated at 9.5, 15-17, and 27°C, respectively. The long-term preferred temperature was determined to be 29-31, 26-27, 23-24, and 14-17°C for acclimation temperatures of 24, 21-22, 17, and 6°C, respectively (Texas Instruments, 1976b). Optimum temperature ranges for the growth of juveniles have been reported as 14-21°C (Davies, 1973; Krouse, 1968) and 15-27°C (Bowker et al., 1969).

Juveniles have been found generally in waters of 0-11 o/oo (Dovel, 1971) or 4-13 o/oo (Clark, 1968). There appears to be some interaction of temperature and salinity on survival or tolerance. For example, juveniles survived abrupt transfers between salt and freshwater at temperatures over a range of 12.8-21.1°C, but were not tolerant of transfers from freshwater to saltwater of 7.2°C within that temperature range (Tagatz, 1961). These tests were performed using mixed salt and freshwater with the resulting pH ranging from 7.4 to 7.6. We have successfully shifted juveniles and yearlings

TABLE 19. ENVIRONMENTAL REQUIREMENTS OF STRIPED BASS JUVENILES AND SUBADULTS

	ABIOTIC FACTORS	
	Survival Range	Optimum Conditions
Temperature	0-30°C	>10 & <25°C
Salinity	0-30 o/oo	10-30 o/oo
Dissolved oxygen	>5% (2.4 mg/l @18°C)	air saturated
Light	no adverse effect	natural photoperiod
Turbidity	0-10 mg/l+; 0-2 g/l*	<4 mg/l+
	BIOTIC FACTORS	
Diet	5-8% body weight (wet) per day	
Density	10 to 2 bass per 100 liters	
Predators	some in natural habitat	
Disease and Parasites	summarized in Table 24	

+ bentonite
* uncontaminated suspended sediments

directly from freshwater to seawater of 28-32 o/oo both at moderate (15-20°C) and low (0-5°C) temperatures. Juveniles (85-105 mm FL) acclimated to 15.6°C in freshwater showed a 50% mortality at 31-34.4°C, while the 50% mortality for those acclimated to 11°C in freshwater was observed at 29.4-30.6°C (Loeber, 1951).

Klyashtorin and Yarzhombek (1975) determined that an increase in salinity up to 10 o/oo produced a short-term increase in oxygen consumption which normalized as the fish adjusted to increased salinity. Meldrim et al. (1974) reported mean resting and active oxygen consumptions at various temperatures and 0, 6 and 12 o/oo salinity. For bass 14.5 to 26.8 cm TL, resting oxygen consumption was 90 mg/kg/hr at 13°C and 209 mg/kg/hr at 20°C at 0 o/oo, and 196 mg/kg/hr at 19°C and 246 mg/kg/hr at 13°C at 6 o/oo. For bass 10.7 to 22.4 cm TL, active oxygen consumption was as high as 1840 mg/kg/hr at 9°C and 12 o/oo and as low as 210 mg/kg/hr at 24°C and 6 o/oo. At 26°C and 0 o/oo the mean active oxygen consumption was reported as 802 mg/kg/hr by these authors.

Striped bass feeding frequency in our flowing seawater system declined sharply below 5°C on natural foods. Adults appear to cease feeding below this temperature, while juveniles continue feeding but on smaller amounts and less often. Feeding of subadults and adults also declined at temperatures over 26°C in seawater. Wawronowicz and Lewis (1979) observed that juveniles on artificial foods (pelleted) ceased feeding in ponds when the temperature fell to 7°C. These bass resumed feeding when the temperature reached 16°C.

The optimum range of dissolved oxygen for juveniles has been given as 6-12 mg/l (Bogdanov et al., 1967), or over 3.6 mg/l (Bowker et al., 1969), or greater than 3 mg/l (Chittenden, 1972; Krouse, 1968).

Dorfman and Westman (1970) observed 80% survival among juvenile bass acclimated to 8.5 mg/l oxygen at 20°C and 6.6 mg/l oxygen at 25.6°C to transfers to 2.0 mg/l oxygen at 20°C and 3.0 mg/l oxygen at 25.6°C, respectively. All of their bass acclimated at 5.9 mg/l oxygen at 32.8°C and transferred to 2.4 mg/l oxygen at 32.8°C died.

Chittenden (1972) found that the effects of handling and salinity on the oxygen requirements of juveniles were negligible or absent for 16 to 18.5°C and 0 o/oo or 10 o/oo. Krouse (1968) observed mortality after 72 hours for juveniles in multivariate experiments of 13, 18 and 25°C, 5, 15, and 25 o/oo (as Instant Ocean), and 1, 3 and 5 mg/l oxygen at pH ranges of 7.1 to 8.7 and constant photoperiod. He found best survival at 18°C and 5 or 15 o/oo with 5 mg/l oxygen levels. He suggested that bass should be able to survive 13 to 25°C and 5 to 25 o/oo water with oxygen levels greater than 3 mg/l. However, at oxygen levels of 1 mg/l or less, complete mortality can be expected. During a thirty day exposure period, striped bass juveniles grew 160% of their initial body weight at average dissolved oxygen concentrations of greater than 7.3 mg/l, but only 130% of their initial body weight at oxygen levels below 3.5 mg/l (Dorfman and Westman, 1970). The average growth rate was given as 0.104 and 0.061 for the high and low oxygen concentrations, respectively.

Peddicord et al. (1975) reported a 240-hour LC50 of 4.6 mg/l bentonite at 18°C and 2 mg/l dissolved oxygen for 50-80 mm bass. Peddicord and McFarland (1978) reported a 10 day LC50 of >4 g/l uncontaminated suspended sediment to juvenile bass at 25 o/oo, 12°C and 8 ppm dissolved oxygen. They estimated a 2 day LC50 of 0.4 g/l contaminated suspended sediments at 25 o/oo, 14°C and 8 ppm dissolved oxygen to juveniles tested. The sediments were collected from the San Francisco Bay area. The uncontaminated sediments tested contained some heavy metals, and were made up of 83% silt, 12% clay and 5% sand. The contaminated sediments contained sulfides, heavy metals, PCBs and DDT, and were composed of 65% silt, 2% clay and 33% sand.

Bowker et al. (1969) observed the tolerance of juvenile bass to a pH range of 6 to 10 in rearing ponds over a range of 22 to 29°C. Tatum et al. (1965) reported a lower lethal pH of 5.3 to juvenile bass during a 24 hours exposure period.

Sazaki et al. (1972) observed 86 to 100% swimming at four minutes exposure to 0.5 to 1.0 fps velocities among 40-50 mm bass. They noted an inverse relationship between survival and impingement velocity in experiments with 25 to 50 mm bass. Kerr (1953) reported that 90-100% of the 25-75 mm bass he tested were swimming at the end of 10 minutes at velocities up to 2.0 fps. Larger bass were able to resist 2 ft/sec velocities for 10 minutes (Kerr, 1953).

Results of toxicological studies on juvenile bass are summarized in Section 12.

Biotic Factors

Striped bass can be classified generally as opportunistic, carnivorous feeders. A great many feeding studies to determine the natural food organisms preferred as well as their relative importance in the diet have been conducted. Some of these are summarized in Table 20 where the relative importance is indicated as percent occurrence in the stomachs of the bass sampled. Young juveniles (<80 mm TL) observed in pond rearing studies (Meshaw, 1969; Humphries, 1971; Harrell et al., 1977) were found to be highly selective in their feeding. These studies determined a high selectivity for *Cyclops* and against *Bosmina* using electivity ratios. As bass mature, general diet preferences become evident. Young bass enter their first fall feeding almost entirely on invertebrates. During the second summer of life they begin feeding on small fish, including young-of-the-year striped bass. In the fall their diet becomes about half fish and half invertebrates, depending on availability. By the third year, especially in spring and summer, their diet becomes almost entirely fish (Markle and Grant, 1970; Manooch, 1973; Stevens, 1966).

A number of feeding studies have been conducted to determine the consumption rate and growth at different temperatures and growing conditions (i.e., ponds, cages, tanks) for a variety of diets, natural and artificial. Much of our study was devoted to feeding studies and some results will be presented in this section in addition to the available

TABLE 20

PREFERRED FOODS OF STRIPED BASS

Author [a]	Length of Bass (mm)	Food Organism	% Frequency of Occurrence in Stomach	Location [b]
Doroshev (1970)	6-9	*Cyclops* nauplii and copepodites	- - -	Moscow (1)
Harper & Jarman (1972)	10-14 TL	Copepods	74	Oklahoma (1)
Humphries (1971)	14-80 TL	Cladocera (Sididea) Copepods (*Cyclops*)	52-58	Virginia (2)
Heubach *et al*. (1963)	5-25	Copepods	41-84	California
Harper *et al*. (1968)	10-39 SL	Copepods (*Diaptomus*)	67-84	Oklahoma (1)
Harper *et al*. (1968)	40-69 SL	Cladocera (*Diaphanosoma*)	82-92	Oklahoma (1)
Heubach *et al*. (1963)	25-76	*Neomysis*	21-86	California
Texas Instruments (1976[c])	0-75	*Gammarus*, Calanoida, *Chironomidae* larvae	-	Hudson River
Townes (1937)	30-110	*Gammarus*	60	Hudson River
Bason (1971)	50-100 FL	*Neomysis*, *Cragon*	35-64, 45	Delaware River
Gomez (1970)	53-100 TL	Diptera (*Chaobrus* & *Chironomus*)	61	Oklahoma (1)
Harper *et al*. (1968)	70-89 SL	Cladocera & Insecta, Fish	71-72, 33	Oklahoma (1)
Heubach *et al*. (1963)	50-115	*Neomysis*, *Corophium*, Copepods	0-83, 11-95, 20-100	California
Texas Instruments (1976[c])	76-125	*Gammarus*, Calanoida	-	Hudson River
Harper & Jarman (1972)	80-109 TL	Cladocera and Copepoda	73-82	Oklahoma (1)
Stevens (1966)	50-230	*Neomysis*	80	California
Goodson (1964)	203-254 FL	Shad	76	California
Ware (1971)	76-350	Shad and fish remains	35-47	Florida (1)
Texas Instruments (1976[c])	116-200	*Microgadus*	-	Hudson River
Thomas (1967)	153-254	*Neomysis*, Fishes	42, 55	California
Manooch (1973)	125-304 TL	Clupeid fish	60	Albemarle Sound
Stevens (1966)	130-350	*Neomysis*	66	California
Shapovalov (1936)	200-490 FL	Fishes, Small Crustaceans	49, 42	California
Thomas (1967)	280-382	Fishes	79	California
Stevens (1966)	260-470	Neomysis, Fishes (*Dorosoma* & *Roccus*)	25, 52	California
Schaefer (1970)	275-399 FL	Mysidacea, Amphipoda	43, 57	Great South Bay
Stevens (1958)	215-763	Clupeoid, Mayfly nymphs	18-100, 0-87	So. Carolina (1)
Schaefer (1970)	400-599 FL	Amphipoda, *Anchoa*	52, 22	Great South Bay
Manooch (1973)	305-714 TL	Clupeid fish	75	Albemarle Sound
Stevens (1966)	380+, 480+	Fishes (*Dorosoma* & *Roccus*)	70	California
Schaefer (1970)	600-940 FL	Amphipoda, Fishes (*Anchoa* & *Urophycis*)	33, 44	Great South Bay
Johnson & Calhoun (1952)	>305	*Neomysis*, *Cragon*, Anchovy	2-20, 35, 11	California
Thomas (1967)	>406	Fishes	95	California
Goodson (1964)	533-863 FL	Shad	99	California
Hollis (1952)	- - -	Fishes (Menhaden, Spot, Croaker)	40-100	Chesapeake Bay

[a] Includes only studies reported where more than 20 fish were examined.

[b] Of study number in parenthesis indicates hatchery source of bass: (1) Moncks Corner, S.C.; (2) Edenton, North Carolina.

information on the diet requirements of juveniles and subadults.

Kelley (1969) compared growth and survival of juvenile striped bass in freshwater troughs (20°C) fed commercial trout diets. He noted conversions of 1.4-2.8 at feeding rates of 3-4% of body weight, with one diet (Purina Trout Chow) yielding greater survival and growth. He concluded that a feeding rate of 3.5% of body weight daily would be appropriate for commercially prepared trout diets, when feeding once a day. Catchings (1973), however, concluded feeding rates of 4 or 5% of body weight per day of trout chow resulted in more efficient food utilization.

Powell (1973) and Valenti *et al*. (1976) reported the results of cage culture feeding studies done in coastal waters of Alabama and New York. respectively. Both found mean conversions of 1.7-4.5 dry feed:live weight for bass groups fed mainly pelleted trout diets. One group receiving ground whole fish-soybean meal diet had a mean conversion of 5.6 (Powell, 1973) when fed 60% per day. We investigated growth in a cage in Rhode Island coastal waters of 50 juveniles fed a diet of ground hake at a rate of 6-12% of their initial live weight (Fig. 29). Their gross efficiency, or conversion, on a wet:wet weight basis, was 27% from June to the July weighing, 32% from July to the August weighing, 21% from August to the September weighing, and 17% from September to the October weighing. Although their gross growth efficiency dropped off during the late August-mid-October period, their growth rate continued to increase at that time (Figure 29).

Redpath (1972) conducted growth studies of juvenile striped bass (5-10 cm) at 8, 12, 16, 20 and 24°C and five feeding levels (1, 3, 5, and 8% of body weight and repletion) on live sludge worms (Tubificidae). Conversion and consumption rates were reported on a dry weight basis. Gross efficiency was lowest at 12° C and increased to 20°C. Maintenance requirements were determined to be 3.37, 21, 7.5 and 11.5 mg/g/day at 8, 12, 16 and 20°C, respectively. He observed higher growth rates over a greater consumption range with greater efficiency at 16°C than at any of the other temperatures studied.

During our study a number of feeding experiments on juvenile and subadults was performed. A number of diets was utilized. Most were readily consumed by the bass - live and frozen brine shrimp, squid, a moist "pellet", menhaden and herring - if presented in a size they could eat. Other diets, especially the dry pellets and some fish, were often not consumed, nor did the bass show any interest in them. Some of the more successful diets and their feeding level estimates are described briefly below.

A feeding study to determine the consumption levels of live brine shrimp post-nauplii and adults by juvenile bass (28-65 mm FL) was performed at 25 o/oo. Two groups were tested at each of three temperatures (18, 21, 24°C). Each group was fed to satiation once a day during three consecutive one week periods. Bass were weighed weekly. A record of the wet weight consumed was maintained for each group. Consumption rate expressed as percent of body weight (wet) per daily meal ranged from 20 to 34% at 24°C, 16 to 30% at 21°C, and 15 to 28% at 18°C. The gross growth efficiency ranged from 2 to 13% at 24°C, 7 to 11% at 21°C, and 6 to 10% at 18°C for these groups.

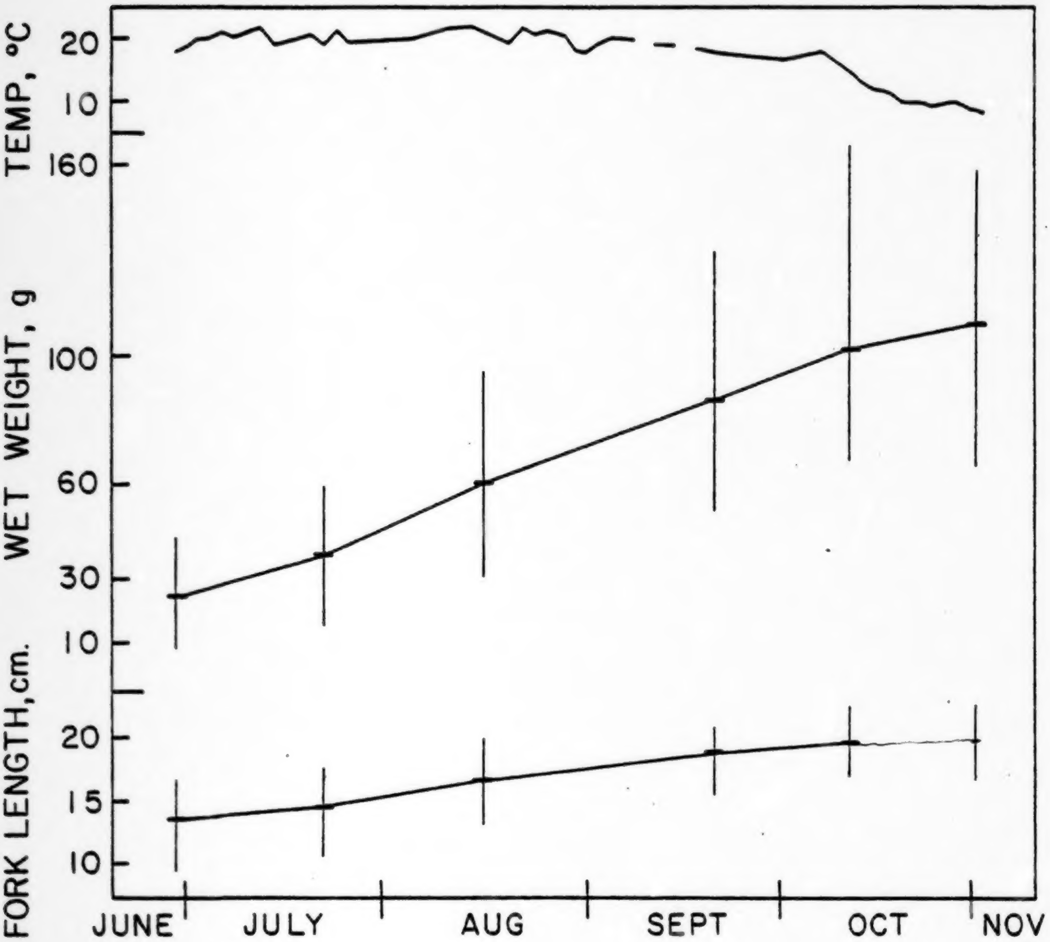


Figure 29. Growth in weight and length for juvenile striped bass held in ambient sea water in a cage. They were fed on a diet of ground hake in addition to the natural prey available in the water column.

During a one week period two individual bass per temperature were observed. Their gross efficiencies when fed to satiation on live brine shrimp daily were 5.9 and 11% at 24°C, 8.2 and 9.3% at 21°C and 4.6 and 5.7% at 18°C.

A study to determine growth and conversion among juveniles (6-10 cm FL) fed cut squid at five levels (2, 4, 8, 12 and 15% body weight wet basis) in groups of 20 bass in ambient filtered seawater was undertaken. The mean growth results are shown in Figure 30 for these five feeding levels. The 2% ration was obviously below the maintenance requirements over the average temperatures of 18 and 20°C for the two periods. Although the 4% level was adequate at 18°C average water temperature, it was about maintenance level at an average temperature of 20°C. The gross growth efficiency for the groups at 18°C were 40, 23, 21 and 19% (wet weight basis) for daily feeding levels of 4, 8, 12 and 15%, respectively. The efficiencies at 20°C were only 3, 21, 27 and 16%, respectively.

A comparison of two diets was conducted on groups of juveniles and yearlings in ambient filtered seawater at 10 and 20°C. The diets were cut squid mantle and a gelatin-squid moist "pellet" (i.e., 48% water, 25% trout crumbles, 12% ground squid and 10% gelatin binder modified from Peterson and Robinson (1967)). Both diets were fed as 0.5 cm square pieces readily eaten at both temperatures. The results are summarized in Table 21 for this study where feeding was ad libitum daily. All of the calculations represented in Table 21 are on a dry weight basis unless otherwise indicated. Percent water was determined (wet-dry weight at 100°C) to be 83.7, 64.4 and 74.6% for the squid, gelatin-squid diets and the striped bass, respectively. Growth was consistently better among the squid-fed bass at the two temperatures as was efficiency on a dry weight basis.

Absorption efficiency and net conversion efficiency were determined for bass fed these diets at 20°C. The efficiencies calculated from the data collected are presented in Table 22. Absorption (A), on a dry weight basis, was calculated by subtracting the amount of feces produced (collected on fine mesh, rinsed and dried) during the period for each diet from the total consumed during the period. This was divided by the amount consumed giving the absorption efficiency as a percent (X 100). The gain during the period divided by the absorption (A) for the period for each diet and expressed as a percent (X 100). These efficiencies were expressed on a caloric basis using mean values of 5350.8, 5552.1, 6574.7, 5762.2 and 4841.5 cal/g (ash-free) for bass, squid, gelatin-squid, squid feces and gelatin-squid feces, respectively, determined during this study. The consistently higher efficiencies for the squid diet over the gelatin-squid diet may be a function of the carbon and nitrogen (protein) in the diets. Carbon:nitrogen analysis on a sample of the diets and feces indicates that the bass utilized more of the nitrogen available in the squid diet (9.5 of 11% N) than in the gelatin-squid diet (5 to 11.5% N). The bass also appeared to utilize more of the carbon available in the squid (28 of 42% C) than of that available in the gelatin (11 of 43% C) formulated diet.

Two groups of subadults were used to determine satiation and feeding levels at ambient sea temperatures. One group was composed of 14 bass which

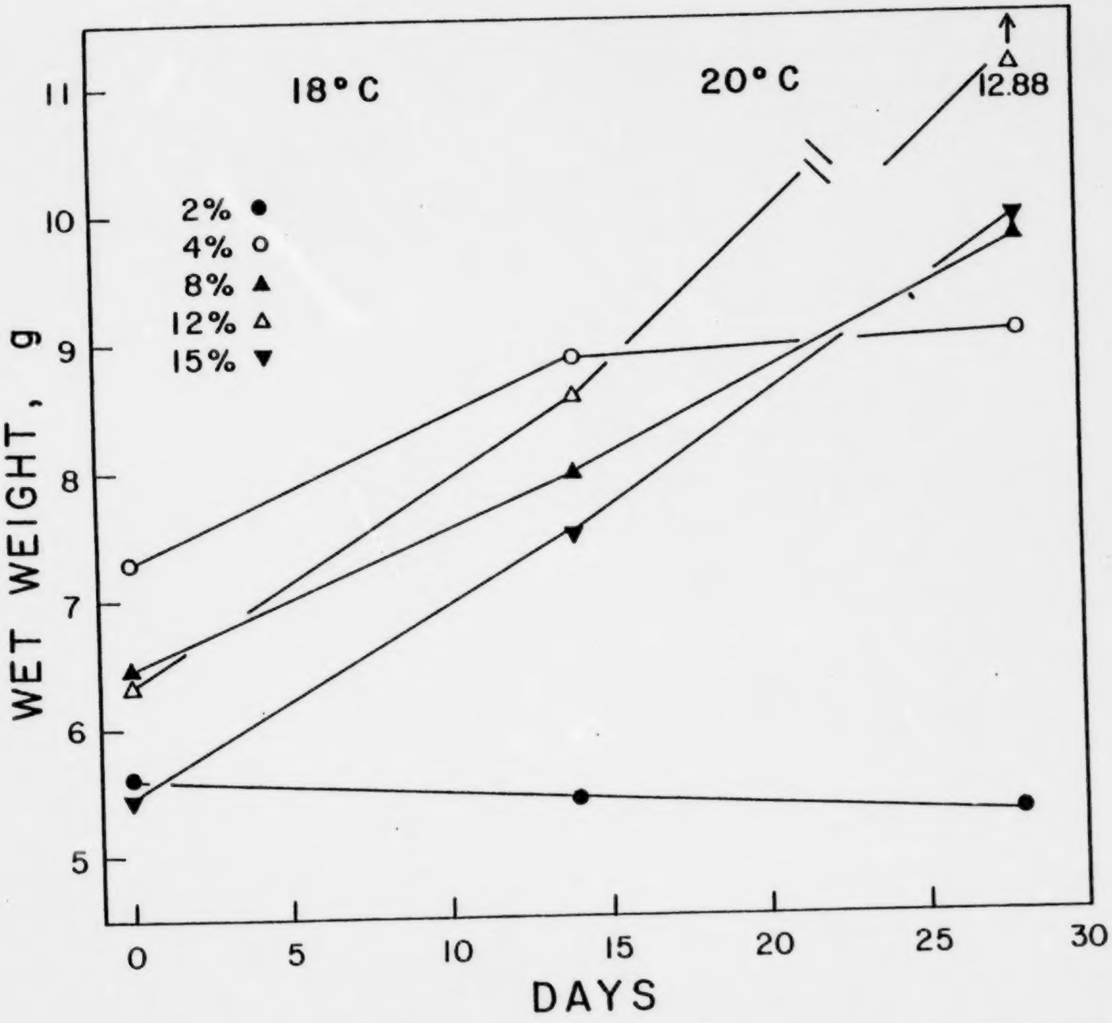


Figure 30. Growth in weight of young-of-the-year striped bass fed at fixed percentages of their live body weight (2 to 15%) per day on cut squid at ambient sea water temperatures of 18 and 20°C.

TABLE 21. SUMMARY OF GROWTH DATA FOR EACH GROUP OF STRIPED BASS FED ONE OF TWO DIETS AT 10 AND 20°C

Diet	Tank	# of Fish	Days in Feeding Period	Mean Dry Weight (gm)		Mean Length (FL cm)	Ration (% body weight daily)	Growth Rate (% daily)	Gross Growth Efficiency (%)	
				Initial	Final				Wet	Dry
20°C Cut Squid	1	18	12	3.10	3.85	10.04	8.1	2.0	16.39	25.0
	2	32	12	7.08	8.62	13.05	6.0	1.8	19.69	30.1
	3	31	12	4.64	5.73	11.36	7.9	2.0	16.24	24.6
	4	18	12	11.20	13.60	15.10	6.2	1.8	19.14	29.0
	5	15	12	19.82	22.85	18.34	4.4	1.3	19.10	28.9
	6	6	12	29.65	35.03	21.26	5.1	1.5	19.73	29.9
20°C Gelatin -Squid	1	28	19	4.20	4.80	10.94	5.9	0.73	18.20	12.2
	2	16	19	8.73	10.17	13.79	5.9	0.86	21.69	14.6
	3	21	19	6.14	8.01	12.26	7.2	1.20	24.63	16.6
	4	9	19	13.30	14.40	15.66	5.3	0.43	12.15	8.2
	5	6	19	20.80	22.80	18.56	5.3	0.50	14.12	9.5
	6	16	19	8.51	9.12	13.66	5.4	0.38	10.55	7.1
	7	9	19	13.90	14.56	16.15	7.9	1.10	20.46	13.8
	8	7	19	25.57	27.01	10.95	4.9	0.73	21.80	14.7
10°C Cut Squid	1	37	18	4.60	4.80	11.40	1.5	0.22	9.70	14.9
	2	16	17	12.40	13.20	15.83	1.8	0.03	1.30	1.9
	3	16	16	11.20	11.80	15.31	1.3	0.32	16.00	24.3
	4	8	16	16.50	17.50	17.38	1.1	0.25	14.50	21.9
10°C Gelatin -Squid	1	27	18	5.80	6.30	12.10	2.2	0.52	36.10	24.2
	2	21	18	9.50	9.60	14.41	1.4	0.03	3.70	2.5
	3	6	18	26.00	25.50	20.16	0.83	-0.12	-21.90	-14.58
	4	27	16	6.30	6.20	12.62	0.65	-0.11	-24.20	-16.28
	5	21	15	9.60	9.50	14.43	0.60	-0.05	-12.40	-8.6
	6	6	15	25.60	25.50	20.18	0.54	-0.02	-5.60	-3.4

TABLE 22. ABSORPTION AND CONVERSION EFFICIENCIES CALCULATED FOR STRIPED BASS FED ONE OF TWO DIETS AT 20°C AND COMPARED TO GROWW GROWTH EFFICIENCY FROM TABLE 21

Diet and Tank	Wet Weight (mean final) g	Gross (growth) Efficiency, % (dry)	Absorption Efficiency, %		Conversion Efficiency (net), %	
			Dry	Caloric (ash-free)	Dry	Caloric (ash-free)
Squid						
1	15.4	25.0	98.2	98.2	25.4	24.5
2	34.5	30.1	98.5	98.4	30.5	29.5
3	22.9	24.6	98.7	98.7	24.9	24.1
4	54.4	29.0	97.9	97.8	29.6	28.6
5	91.4	28.9	97.7	97.6	29.6	28.6
6	140.2	29.9	97.1	96.9	30.9	29.3
Gelatin-squid						
1	19.2	12.2	86.5	90.0	14.0	11.0
2	40.7	14.6	84.7	88.7	17.2	13.4
3	30.1	16.6	87.5	90.8	18.9	14.8
4	57.5	8.2	87.4	90.7	9.4	7.4
5	91.2	9.5	88.1	91.3	10.8	8.5

ranged initially from 32.4 to 39.5 cm FL and 410-797 g. The second group consisted of 20 bass ranging in size from 22.5 to 27.5 cm FL and 151.5-285.5 g. Each was weighed from a stock pool into a clean 12 foot pool with flowing seawater in August (Figure 31). Cut frozen squid became the diet, since all was consumed at the time of first feeding. The first group of 14 during an initial three weeks (to September) had an efficiency of 7% on a wet weight basis. Both groups became satiated at 4-8% of their last weight when fed daily. This rose to 12% following one day's fast. Food consumption, when presented at more than one feeding a day, declined with each feeding. Given satiation feedings at two hour intervals, the first was about 60%, the second about 25%, and third about 16% of the day's total consumption. Over a six week period the 20 bass group (to October) had an 18% food conversion on a wet weight basis. During a five week feeding period from September to October, the 14 bass group had a 21% food conversion. These observations were at average ambient sea temperatures shown in Figure 31. In an eight week feeding period, from October to December, in which the water temperature fell from 16 to 5°C (mean 11.2°C) the group of 14 bass had an efficiency of 17%. The satiation level of this group declined to an average 2.9% of their live body weight. This level is about one-quarter to one-half of the satiation level observed during the two prior periods. The effects of temperature on feeding levels are obvious from Table 21 and Figures 30 and 31.

The density of 10-2 fish per 100 l given in Table 19 is based primarily on laboratory data rather than on field data where confinement is not a problem. If the 10 bass are juveniles of 10 g live weight each, the optimum density of 1 g per liter would be achieved. If the bass are yearlings of 140 g each, then the density would be 3 g per liter. We found this satisfactory for feeding activity and growth in flowing seawater or well aerated recirculating systems. It was the greatest density of any of our groups for which diet and feeding, oxygen consumption and ammonia excretion data presented were collected. Under culture conditions, density is important especially when the activity demands of the fish are added to the effects of normal respiration on the water quality. Observations of changes in respiration and excretion offer indications of activity and can, thereby, yield a more specific determination of optimum density.

Studies on groups of juvenile and yearling bass in filtered ambient seawater showed increased mean oxygen consumption with temperature and feeding (Figs. 32 and 33). The number of bass per group ranged from 37 to 4 with mean group weights of 6 to 135 grams live weight. The effect of rearing a 40 g bass at 20°C rather than 8°C is an average increase of at least 1.5 times in the oxygen needed for routine metabolism (Fig. 32). At 20°C the average oxygen consumption increased 2.5 times with feeding, but only increased 1.3 times at 12°C with feeding (Figure 33). Yarzhombek and Klysahtorin (1974) determined by comparison of means that metabolism with standard activity was 1.3 times the calculated resting metabolism. Kruger and Brocksen (1978) calculated the relationship between swimming speed and oxygen consumption at three swimming velocities and five temperatures for 22-68 g bass. The relationship varied from $\log Y = 0.6452 + 0.0345X$ at 8°C

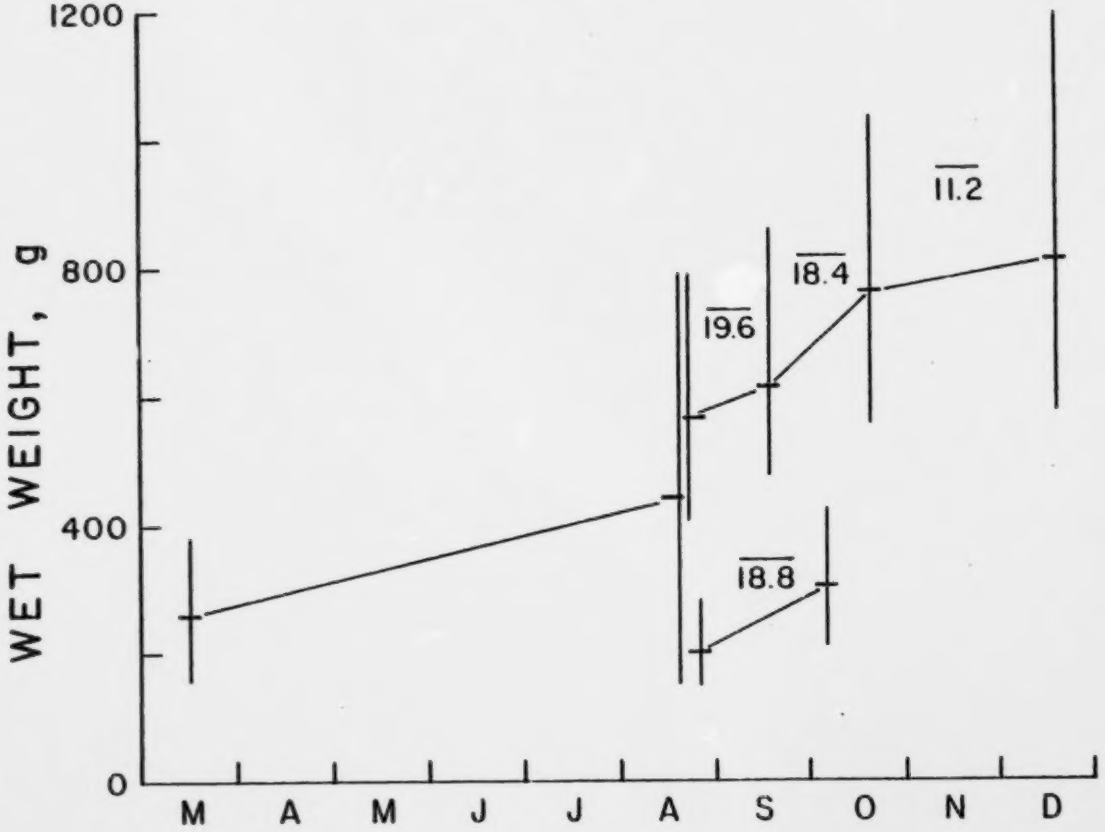


Figure 31. Growth in weight of subadult striped bass fed to satiation daily in ambient seawater.

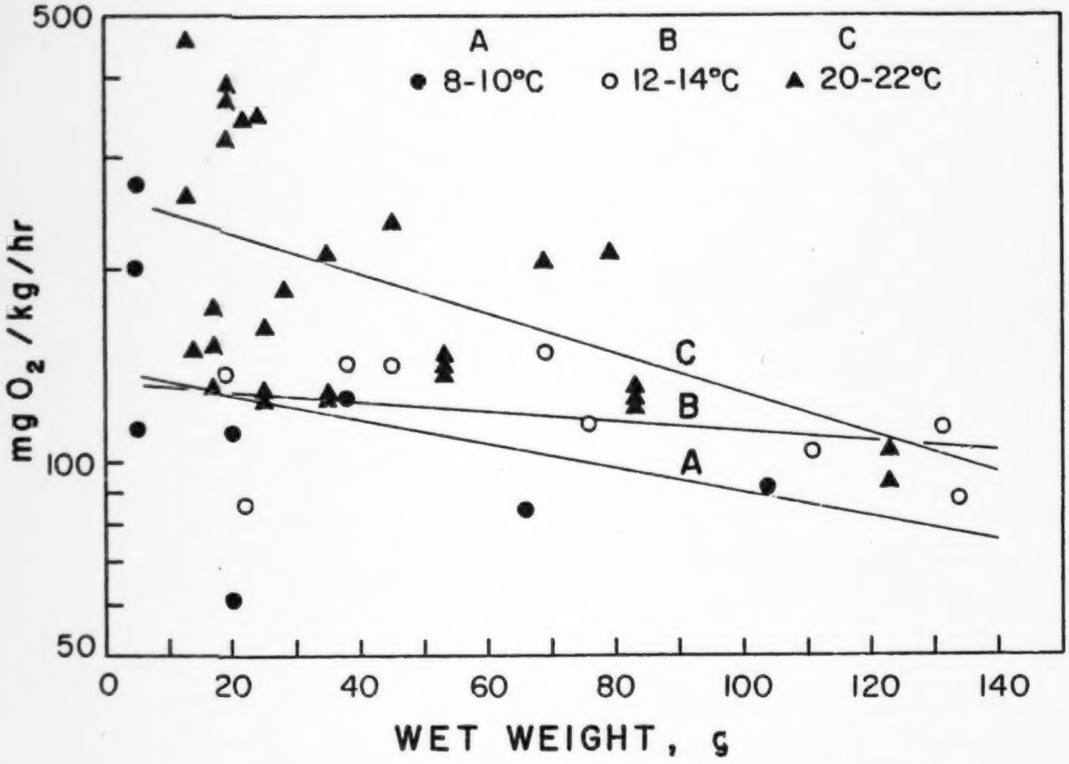


Figure 32. Routine oxygen consumption (y = mg O_2/kg, wet weight/hr) determined for juvenile and yearling striped bass of wet weight (x) over three temperature ranges (A, B and C). Regression lines calculated for the data shown are

A: $\log_{10} y = -0.0008 x + 2.11$ ($r = -0.58$);
B: $\log_{10} y = -0.0008 x + 2.13$ ($r = -0.43$);
C: $\log_{10} y = -0.003 x + 2.42$ ($r = -0.59$).

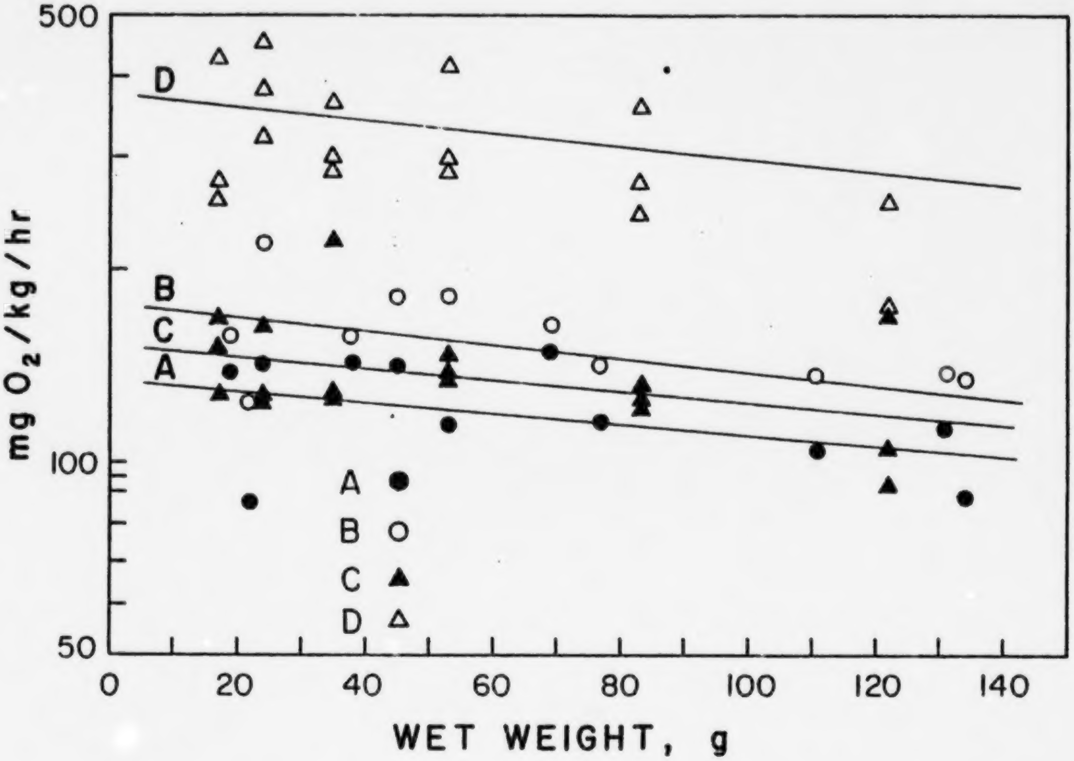


Figure 33. Routine oxygen consumption (y = mg O_2/kg, wet weight/hr) determined for juvenile and yearling striped bass of wet weight (x) before (A) and after (B) feeding over the temperature range of 12 to 14°C and before (C) and after (D) feeding at temperatures of 20 to 22°C. Regression equations calculated from the data shown are

A: $\log_{10} y = -0.0008 x + 2.13$ $(r = -0.43)$;
B: $\log_{10} y = -0.0008 x + 2.24$ $(r = -0.47)$;
C: $\log_{10} y = -0.0009 x + 2.18$ $(r = -0.42)$;
D: $\log_{10} y = -0.001 x + 2.58$ $(r = -0.58)$.

to log Y = 1.1778 + 0.0112X at $16^{o}C$ to log Y = 1.3322 + 0.0211 at $24^{o}C$ where Y = oxygen consumption in mg/g/hr and X = swimming velocity in cm/sec.

The excretion rates of juvenile and yearling bass are influenced by temperature, feeding and salinity. Figure 34 indicates relationships between temperature and feeding activity on ammonia excretion rates. These results are from the group studies shown in Figures 32 and 33. The greatest effect of feeding appears to be on the smallest juveniles (10 g) at the higher temperatures. The general increase for either temperature for an average 40 g bass is at least 1.5 times the rate prior to feeding. Table 23 summarizes individual ammonia excretion determinations made during this study at a series of temperatures and salinities for juvenile and subadult bass. There appears to be a reduction in excretion rate with increasing salinity. It was coupled with a marked increase in survival, especially among juveniles (Fig. 35). This leads to the suggestion that juvenile and yearling bass can probably be kept at fairly high densities if the salinity is raised above that of freshwater. Figure 35 also indicates that bass are tolerant to relatively high ammonia concentrations in freshwater (2 ppm) and very high ammonia concentrations in seawater (8 ppm).

In nature predators of juveniles, other than larger striped bass, include tomcod, bluefish, and other piscivors. The subadults are not often prey except to man.

Table 24 lists the parasites and observed site of infection in striped bass including two parasites (Cryptocaryon sp. and Myxosporidin sp.) not previously reported, but observed during this study. Infections are usually not intense enough to cause mortality among wild populations unless other stresses are present. However, under culture conditions diseases and parasites may become epidemic. Controls for parasites are described later in this section.

The most common abnormality reported for striped bass is "pugheadness" (Alperin, 1965; Cheek, 1966; Grinstead, 1971; Gruder, 1930; Lyman, 1961; Mansueti, 1960; Talbot, 1967). It has been noted among juveniles and subadults. The osteology has been described and its causes discussed. It is generally thought to be caused by environment and/or heredity, rather than mechanical damage during development.

CULTURE METHODOLOGY

Capture Methods

Juveniles can be reared from larvae or caught easily using a beach seine (100 ft. 1/4" mesh) from shoal waters. Other methods include an epibenthic sled, tucker trawl, bottom trawl, balloon trawl (Trent, 1962), otter trawl (Saski, 1966), or a tow net on skis (Turner and Chadwick, 1972). Yearlings are often caught in beach seines and bottom or otter trawls. Larger subadults are generally caught in haul seines, small mesh gill nets, fish traps, and even by handlines. Capture methods for pond culture are described by Bonn et al. (1976) for properly constructed rearing ponds. The

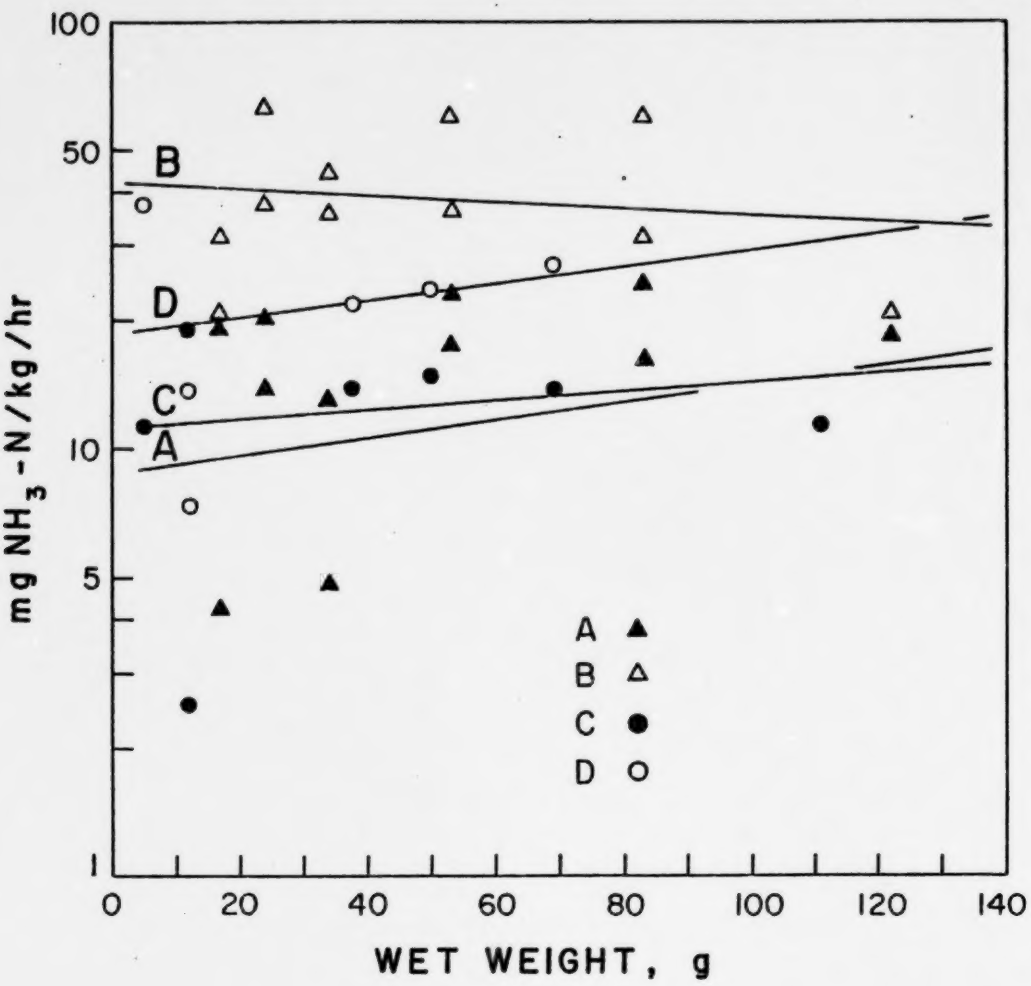


Figure 34. Ammonia excretion rate (y = mg NH_3-N/kg, wet weight/hr) determined for juvenile and yearling striped bass of wet weight (x) before (A) and after (B) feeding at 18 to 22°C and before (C) and after (D) feeding at 8 to 12°C. The regression equations calculated from the data shown are

A: $\log_{10} y = 0.002 x + 0.947$ ($r = 0.28$);
B: $\log_{10} y = -0.007 x + 1.612$ ($r = 0.16$);
C: $\log_{10} y = 0.001 x + 1.045$ ($r = 0.21$);
D: $\log_{10} y = 0.002 x + 1.265$ ($r = 0.28$).

TABLE 23. EXCRETION RATES MEASURED FORTY-EIGHT HOURS AFTER LAST MEAL FOR INDIVIDUAL JUVENILE AND SUBADULT STRIPED BASS

Wet weight (g)	Number per test	Temperature (°C)	Salinity (°/oo)	Ammonia excretion ($N-NH_3$ mg/kg/day)
2.5-6.9	10	20	0	864.6 ± 189
2.7-5.7	9	20	30	637.8 ± 161
3.6-5.3	10	6	30	104.4 ± 33
3.6-7.2	6	6	0	138.6 ± 35
3.6-7.2	4	10	10	122.5 ± 56
45	1	6	0	76.6
75	1	6	0	63.6
44	1	10	10	51.4
72	1	10	10	38.4
74	1	16	30	1005.4
176	1	16	30	519.8
229	1	16	30	323.6
244	1	16	30	351.4
305	1	16	30	359.5

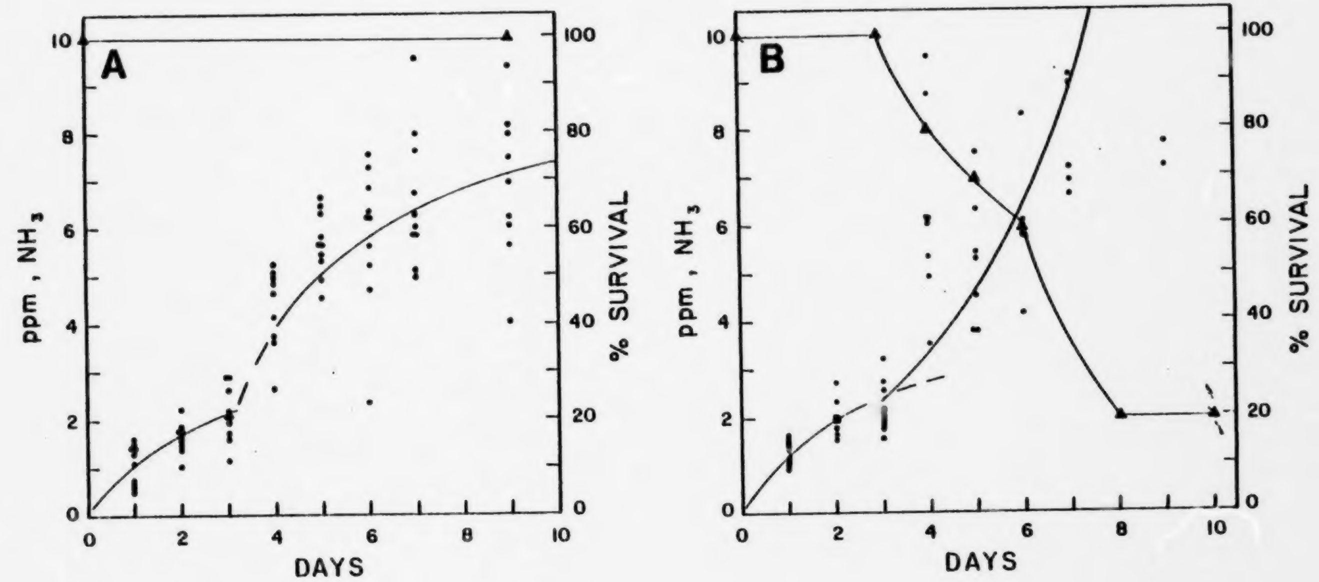


Figure 35. Cumulative ammonia excretion (dots) by individual juvenile striped bass in seawater (A) and freshwater (B). Percent survival (triangles) of all individuals in seawater (A) and freshwater (B) is also shown.

TABLE 24. PARASITES AND DISEASES OF STRIPED BASS REPORTED FROM THE LITERATURE AND THIS STUDY

Causitive Agent	Site of Infection	Authors
Virus		
Lymphocystis	general	Krantz (1970), Paperna & Zwerner (1976b)
	dermal epidermis	Wolke (1975)
Bedsonia - Psittacosis group		
Epithelocystis	gills	Wolke *et al*. (1970); Paperna & Zwerner (1976b)
Bacteria		
Aeromonas, *Vibrio* and *Pseudomonas* spp.	fins	Mahoney *et al*. (1973)
Aeromonas hydrophilia	fins, kidney	Hawke (1976)
Chondrococcus columnaris	external, kidney	Bowker *et al*. (1969); Reeves (1972)
Enterobacter cloaeae	fins, kidney	Hawke (1976)
Flexibacteria (Chondrococcus) *columnaris*	caudal peduncle	Hawke (1976)
Myxobacteria sp.	systemic	Allen (1972)
Pasteurella sp.	systemic	Snieskzo *et al*. (1964)
Pasteurella piscicida	viscera systemic	Paperna and Zwerner (1976b)
Vibrio anguillarum	systemic	Hawke (1976)
Fungus		
Branchiomyces sp.	gill	Meyer and Robinson (1973)
Saprolegnia	fin	Hawke (1976)
fungus and bacteria	air bladder	Wales (1946)
Protozoan		
Ambiphyra sp.	gills	Hawke (1976)
Bodomonas sp.	gills	Hawke (1976)
Chilodonella sp.	gills	Hawke (1976)
Colponema sp.	gills	Paperna & Zwerner (1976b); Texas Instruments (1977a)
Costia sp.	gills	Hawke (1976)
Cryptocarynosis	gills	This study
Epistylis sp.	gills	Paperna & Zwerner (1976b)
Glossatella sp.	gills	Reeve (1972); Paperna & Zwerner (1976b)
Ichthyophthirius multifilis	gills	Texas Instruments (1977a)
Kudoa cerebralis	brain	Paperna & Zwerner (1974 and 1976b)
Myxosoma morone sp.	cartilage, bones	Paperna & Zwerner (1976b)
Myxosporidiosis	brain	This study
Nosema sp.	gills	Paperna & Zwerner (1976b)
Oodinium sp.	gills	Paperna & Zwerner (1976b)
Trichodina sp.	gills	Bowker *et al*. (1969); Reeves (1972); Wolke (1975)
T. davisi	gills	Paperna & Zwerner (1976b)
	gills and skin	Hawke (1976)
Trichodinella sp. and *Paratrichodina* sp.	gills	Hawke (1976)
Trichodinella sp. and *Scyphidia* sp.	gills; gills & skin/gills	Hawke (1975); Paperna & Zwerner (1976b)
Trichophyra sp.	gills	Bowker *et al*. (1969); Hawke (1976); Paperna & Zwerner (1976b)
Tripartiella sp.	gills	Hawke (1976)
Tremadodes		
- Monogenetic		
Ancyrocephaline unidentified	gills	Paperna & Zwerner (1976b)
Aristocleidus hastatus	-	Merriman (1941)
Gyrodactylus sp.	gills	Paperna & Zwerner (1976b)
Lepidotes collinsi	-	Merriman (1941)
Microcotyle macrura	gills	Paperna & Zwerner (1976b) Merriman (1941)
Microcotyle acanthophallus	-	Merriman (1941)
Microcotyle eneides		Merriman (1941)

(continued)

TABLE 24 (continued)

Causitive Agent	Site of Infection	Authors
- Digenetic		
Distomum tornatum	intestine	Linton (1901)
D. tenue	rectum	Linton (1898)
(*Stephanostomum tenue*)	intestine	Paperna & Zwerner (1976b)
D. glactosomum	-	Merriman (1941)
Lepocreadium setiferoides	intestine	Paperna & Zwerner (1976b)
L. areolatum	intestine	Paperna & Zwerner (1976b)
L. californianum	intestine	Edwards & Nahhus (1968)
Neochasmus sogandaresi	intestine & pyloric caeca	Overstreet (1971)
Metacercaria		
Ascocotylid type sp.	viscera	Paperna & Zwerner (1976b)
Clinostomum marginatum	connective tissue & muscle	Paperna & Zwerner (1976b)
Diplostomulum flexidandum	eyelens	Bowker *et al*. (1969)
Diplostomulum sp.	spleen	Paperna & Zwerner (1976b)
Neascus sp.	skin	Paperna & Zwerner (1976b)
Posthodiplostomum minimum	viscera	Bowker *et al*. (1969)
Cestodes		
Proteocephalid larvae - type A	mesenteries	Paperna & Zwerner (1976b)
Proteocephalid larvae - type B	mesenteries & liver	Paperna & Zwerner (1976b)
Rhynchobothrium speciosum	cysts in viscera	Linton (1901)
R. bulbifer	pleroceri in intestine	Linton (1924)
Scolex pleuronectis	intestine	Paperna & Zwerner (1976b)
Trypanorhynchid pleurocercoid	mesenteries	Paperna & Zwerner (1976b)
Acanthocephala		
Echinorhynchus proteus	rectum	Linton (1901)
E. acus (*E. gadi*)	-	Linton (1901)
Pomphorhynchus rocci	pertoneal of intestine	Cordonnier & Ward (1967)
	large intestine	Johnson & Harkema (1970)
	hindgut	Wolke (1975)
	intestine	Paperna & Zwerner (1976b)
	viscera, larvae	Paperna & Zwerner (1976b)
Nematoda		
Ascaris sp.	immature stomach	Linton (1901)
Cuculanus sp.	intestine	Paperna & Zwerner (1976b)
Filaria rubia (*Dicheilonema rubryni*)	flesh, peritoneum	Linton (1901); Merriman (1941)
Goezia sp.	stomach	Gaines & Rogers (1972)
	intestine	Paperna & Zwerner (1976b)
Lecanocephalus annulatus	peritoneum	Linton (1901)
(*Goezia annulata*)	stomach mucose	Merriman (1941)
Philometra rubra (Filaria)	visceral cavity & mesenteries	Paperna & Zwerner (1976b)
Spinithectus sp. (larva)	intestine wall	Paperna & Zwerner (1976b)
Crustacea		
Achtheres lacae	ectoparasite	Merriman (1941)
Aegathoa cf. *oculata*	skin	Paperna & Zwerner (1976b)
Argulus alosae	ectoparasite	Merriman (1941)
Argulus bicolor	skin & gills	Paperna & Zwerner (1976b)
Caligus rapax	ectoparasite	Merriman (1941)
Caligus sp.	gills	Paperna & Zwerner (1976b)
Ergasilus labracis	ectoparasite	Leidy (1888); Merriman (1941)
	gills	Paperna & Zwerner (1976a & b); Hogan & Williams (1976)
Ergasilus cf. *lizae*	gills	Paperna & Zwerner (1976b)
Livoneca avalis	mouth & gill	Alperin (1966b);Lindsay & Moran (1976)
	4th gill arch	Paperna & Zwerner (1976b)
Hirudinea		
Myzobdella lugubris	skin	Paperna & Zwerner (1976b)
Mollusca		
Glochidia	gills	Paperna & Zwerner (1976b)
Vertebrata		
Rissola marginata	coelom	Hamer (1966)
Neoplasm		
Nephroblastoma	renal	Helmboldt & Wyand (1971)

best methods are those which stress the fish the least.

Post-Capture Handling

It is generally good practice to keep any newly captured bass separate from groups already under culture. Mixing, especially without any prophylaxis, often results in loss of all due to infection of one fish. Most juveniles and yearlings are captured in fresh waters. Bonn et al. (1976) suggest holding pond harvested fingerling bass for 24 hours and treating with Furacin at 100-500 ppm with 1% salt for 2 to 5 hour periods prior to shipping. This could probably be done for estuarine caught bass after transporting them to their destination. Also effective, as we have found, is a malachite green bath (1 mg/l) or a formalin bath (0.25 ml 37%/l). Further discussion of disease control is presented at the end of this section.

Transportation

Bonn et al. (1976) describe hauling pond harvested juveniles in tank trucks with 1% salt (NaCl) and 21 ppm MS-222, or 0.35 ppm Quinaldine, to reduce activity. They suggest hauling densities of 1/4 to 1/2 pound per gallon (28-44 g/l) with good aeration. We have had fewer latent mortalities when transporting juveniles in 10-15 o/oo salinity than in freshwater (see Fig. 36). The 10-15 o/oo salinity was made up from filtered seawater and river water with a gentle stream of oxygen as aeration in the transport containers. Our transport containers were polyethylene drums, or plastic garbage cans, with heavy clear plastic (4-6 mil) bag liners. These were filled about half (20-25 gallons) with the transport water and 50-100 juveniles. An oxygen line was run into the bag and the top tied off around it. Thus, not only was the water fully saturated with oxygen, but also the space above it. If it became warm, ice chunks were placed along the top of the inflated bag and allowed to melt down the sides into the bottom of the drum or can. This kept water temperatures below 16-18°C even during the hottest collection times.

Recovery Period

Striped bass juveniles and subadults recover from transportation quite rapidly. At temperatures of 16-20°C we found they eat some food as soon as the next day after capture and transportation. In any case, some food should be presented each day after arrival to be sure they have food as soon as they are recovered. We found better transition to the culture system when the bass transported at 10-15 o/oo were kept at that salinity for a few days till feeding initiated before reducing or increasing the salinity to that of their permanent culture water. The smaller bass adapt very quickly whereas the larger ones require a few days longer to fully make the transition to the culture system.

Handling Procedures

Following complete metamorphosis, juveniles can be handled with nets. They should not be kept out of the water any longer than necessary. Dipping

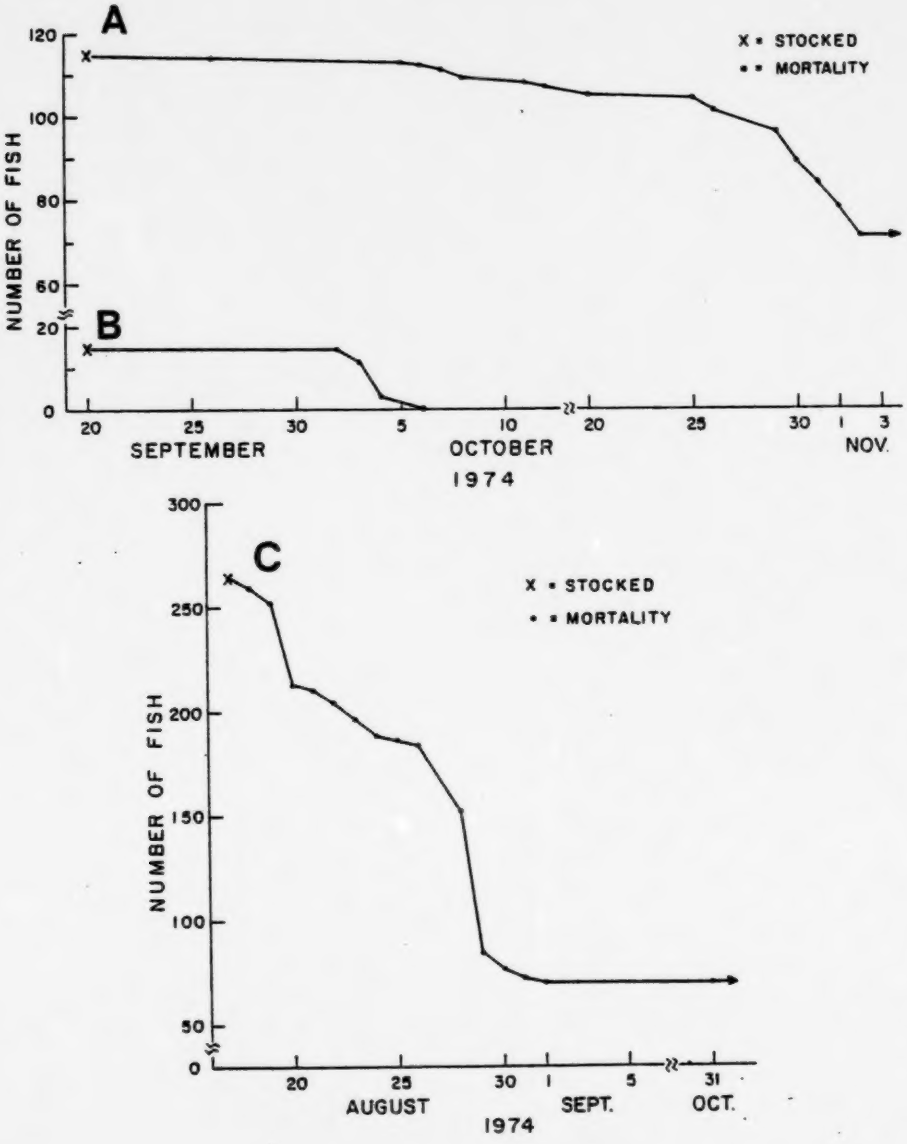


Figure 36. Holding mortality of juvenile striped bass seined from rivers in Maryland and New York either (A) transported and held in 10°/oo water, or (B) transported and held in freshwater, or (C) transported and held in freshwater until transferred to seawater about August 30th.

a fish out of a tank is often made easier by using some crowding device. Juveniles and subadults are capable of rapid acceleration to avoid netting, and crowding helps to avoid wearing the fish out chasing it around the tank. In addition, it speeds up the process of transfering the fish.

Bass of this size are very easy to observe as they swim about the tanks. If they are not excited or active, they can be counted or watched during routine swimming in the tank. This is also an easy way to observe if there are any sick or stressed fish in the system. These will usually swim slower and/or appear darker and/or swim nearer the water's surface than the rest of the bass.

Subadult and juvenile bass are easily weighed or lengthed. However, they should be anesthetized during this process to prevent undue stress. We have found either MS-222 or quinaldine to be very effective anesthetics. These are made up in rearing water. It takes about 1-2 minutes to 'knock the fish out' (quinaldine is slightly faster than MS-222), and about 5-10 minutes for the fish to fully recover. The dosages of these anesthetics that were found most effective were 0.3 g/l MS-222 or 0.025 ml/l quinaldine. Other anesthetics can be used. A very good review of anesthetics, their dosages and uses for fish is provided in McFarland and Klontz (1969).

Maintenance Procedures

Culture Vessels--

Culture vessels for juveniles and older bass can be any shape or size provided the fish are not crowded and the construction material is not toxic. Square or round tanks, raceways or troughs, and swimming pools have all been used. Open flow systems using well water or seawater and recirculating systems with various filter types have been used for juveniles following metamorphosis. (Descriptions of some are in Section 9.)

Stocking Density--

Factors influencing density have been discussed earlier in the biotic requirement section. In an open flow culture system, the densities can probably be maintained on the high side (>1 g/l). However, in recirculating systems it is probably wise not to overload the filter, especially if it is a new system. In either event the dissolved oxygen in the culture water must ensure at least 100 mg O_2 per kilogram of fish hourly (Figs. 32 and 33). The density recommended is 1-3 g per liter (see Table 19).

Maintaining Water Quality--

In a flow through, or once-used, system water quality is maintained more easily than in a recirculating system. However, monitoring of dissolved oxygen, temperature, salinity, and pH should be done routinely and ammonia concentrations frequently. In recirculating systems all of these are necessary, and nitrate-nitrite determinations should be included in the monitoring procedures. Uneaten food and fecal material should be removed from the culture tanks at least daily to prevent an additional source of biological oxygen demand and possible "culture media" for potential disease organisms.

Diet--

The caloric and percent composition for some of the foods of striped bass juveniles are available from the literature and this study (Table 25). It appears that their caloric requirements are similar to their caloric content and they utilize diets higher in protein than fats. We observed (Table 22) that juveniles and yearlings absorbed 97% of the squid consumed (on a dry weight basis) of which 28% was converted (on a dry weight basis) at 20°C (see Table 21 for ration and growth of these bass). Thus, 27% of the dry weight of this diet consumed was available for metabolism, growth, and activity of these fish.

Feeding rate depends to some extent on the digestion time of the bass. We used a "bead-tagging" method to determine the evacuation time for food items fed to juveniles and yearlings at three average temperatures. Figure 37 shows that evacuation time estimated from these studies was affected by fish size, temperature and the ad libitum ration consumed. The size of the ration was influenced by fish size and temperature as seen in Table 21. From our studies it appears that juvenile and yearling bass digest most of a given ration within two to four days at 14-16°C, but need six to nine days for digestion at 6°C. We also observed ad libitum consumption rates at a number of deprivation times at 18-20°C for juvenile and yearling bass. Juveniles fed cut squid consumed the greatest percent of their body weight at one feeding after 20-25 hours deprivation. Yearlings fed squid consumed the greatest amount after 15-20 hours since their last feeding. Yearlings fed commercial trout pellets (3/16"), however, consumed the maximum percentage body weight after only about 10 hours deprivation. In each case the maximum amount consumed did not increase substantially from the percentage consumed at the above times when food was withheld for up to 65 hours. Our data indicate feeding should be daily at temperatures above about 14°C, alternate to every third day at temperatures of 5 to 10°C, and weekly for temperatures less than 5°C. At 4-5°C, bass generally do not feed actively. This regime is recommended for naturally derived foods (e.g., squid or fish) but not for pellets. Pellets should be presented more frequently since consumption rate per feeding appears smaller and digestion time may be faster than for natural foods. We were able to utilize pellets as a satisfactory diet (i.e., were eaten and promoted growth) only at temperatures in excess of 18-20°C.

Juveniles (5-12 cm FL) could easily consume food items of up to 3 mm diameter, but preferred those 2 mm in diameter. Yearlings (13-16 cm FL) were observed to consume food particles up to 8 mm diameter, but preferred those 2-4 mm. Yearlings+ (18-23 cm FL) consumed items up to 12 mm diameter, but more often consumed those 6-8 mm in diameter. The food particles tested were discs cut from frozen squid mantle with a cork borer set. The mantle was about 1-2 mm thick. All bass usually consumed the smaller sizes when mixed sizes were offered.

Growth of juveniles and yearlings fed ad libitum on two diets that were acceptable over a temperature range is shown in Figure 38. It appears that a diet of squid provided a slightly more uniform and consistently increasing growth rate than did the moist gelatin-squid-trout starter diet. The average temperatures for the periods was 20 (Fig. 38A), 16 (Fig. 38B to date 40), and

TABLE 25. CALORIES AND PERCENT COMPOSITION OF SOME FOODS OF JUVENILES AND SUBADULT STRIPED BASS

Prey	Calories per gram (dry weight)	Percent Composition (dry weight) Lipid	Protein	% Water
Gammarus sp.	6737 ± 863 (4)*	0.8	6.4 (1)	
Neomysis integer (2)	--	13.0	70.9	
Callinectes sapidus (3)	81.5 cal/100g	1.0	16.1 g/100g	81.2
Clupea harengus (3)	215 cal/100g (wet weight)	15.7	18.2 g/100g	60.1
(5)	6411			68.8
Squid (3)	89 cal/100g	1.0	15.3 g/100g	79.3
(6)	5552 ± 274	-	68.8**	83.7

* Sources: (1) Phillips *et al*., 1954; (2) Raymont *et al*., 1963; (3) Sidwell *et al*. 1974; (4) Slobodkin and Richman, 1961; (5) Pandian, 1970; (6) This Study.

** Calculated from percent nitrogen (i.e., 11% N x 6.25).

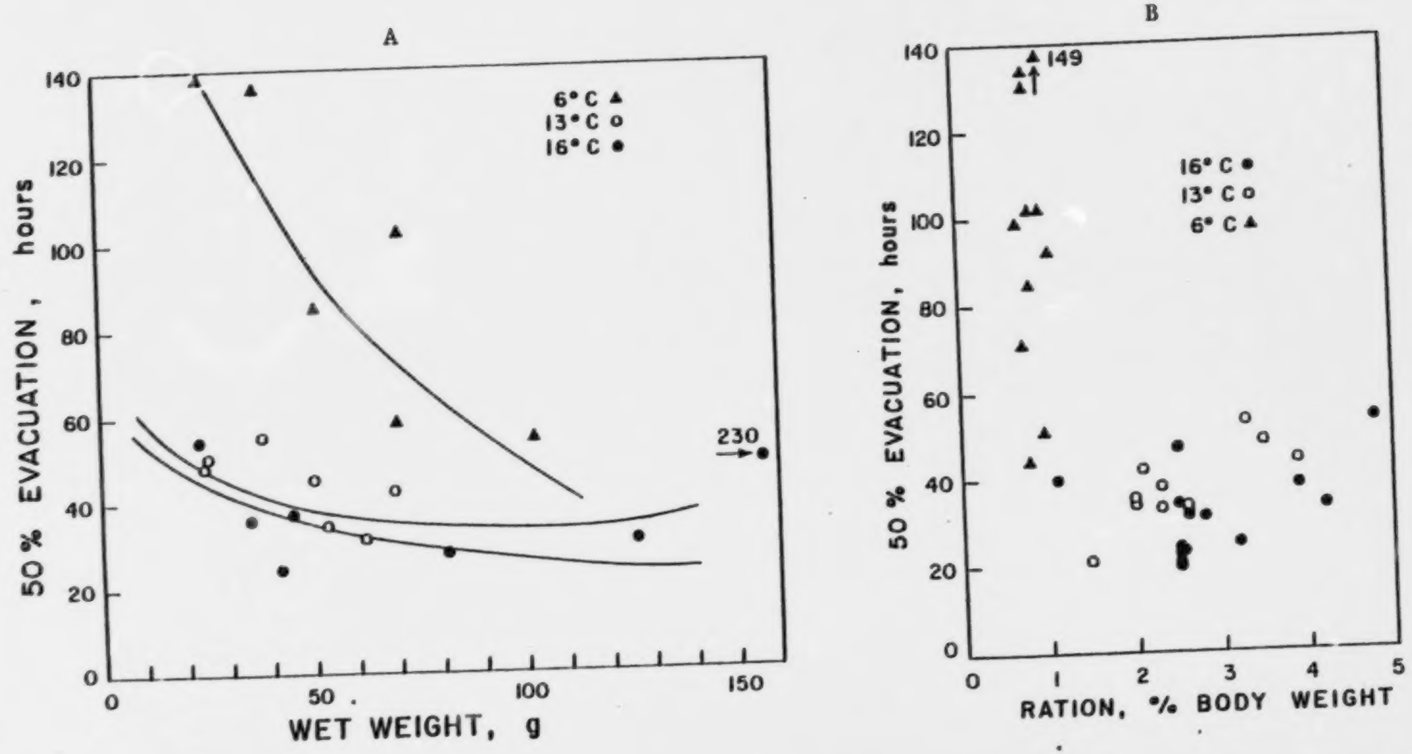


Figure 37. A. Plot of probit estimated time (hours) for 50% evacuation for bass of mean wet weight (g) per group (Table 21) for average temperatures indicated. Lines are the best fit (based on the correlation coefficient) from graphic (13°C) and probit (6 and 16°C) estimations from data.

B. Plot of graphically estimated time (hours) for 50% evacuation of ration, as a percent of live body weight, consumed by each group.

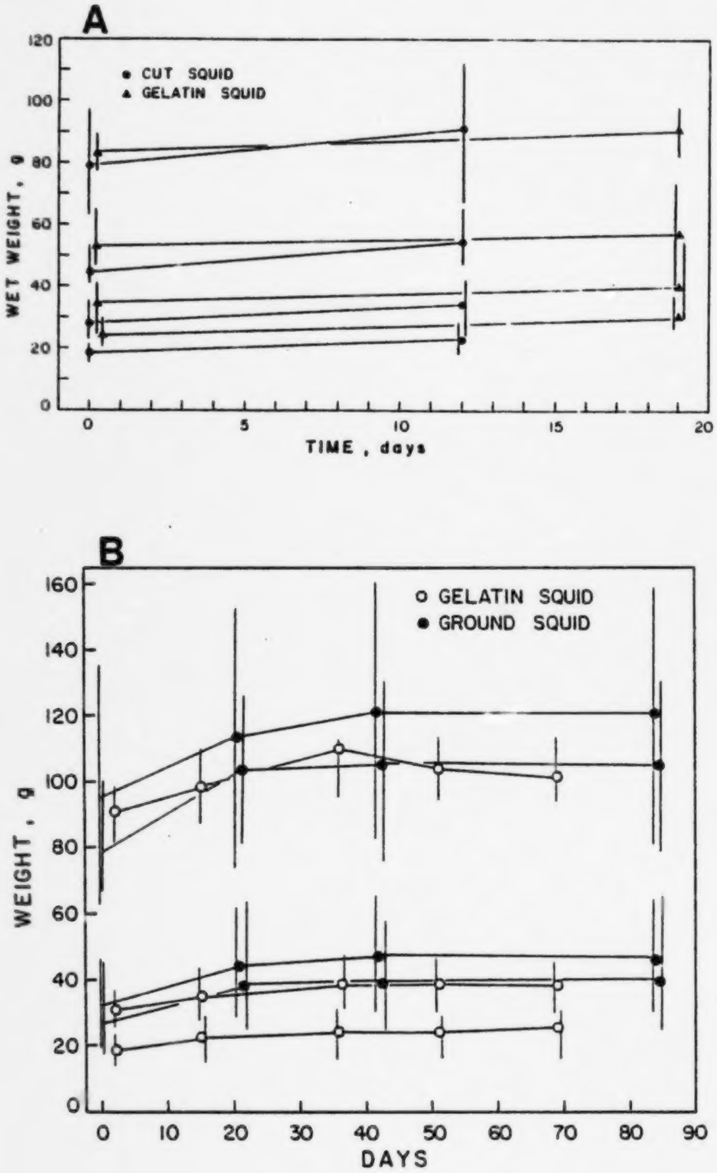


Figure 38. Growth in weight of juvenile and yearling striped bass fed daily on one of three diets in sea water at average temperatures of 20°C (A) and 16 and 12°C (B).

12°C (Figure 38B after day 40).

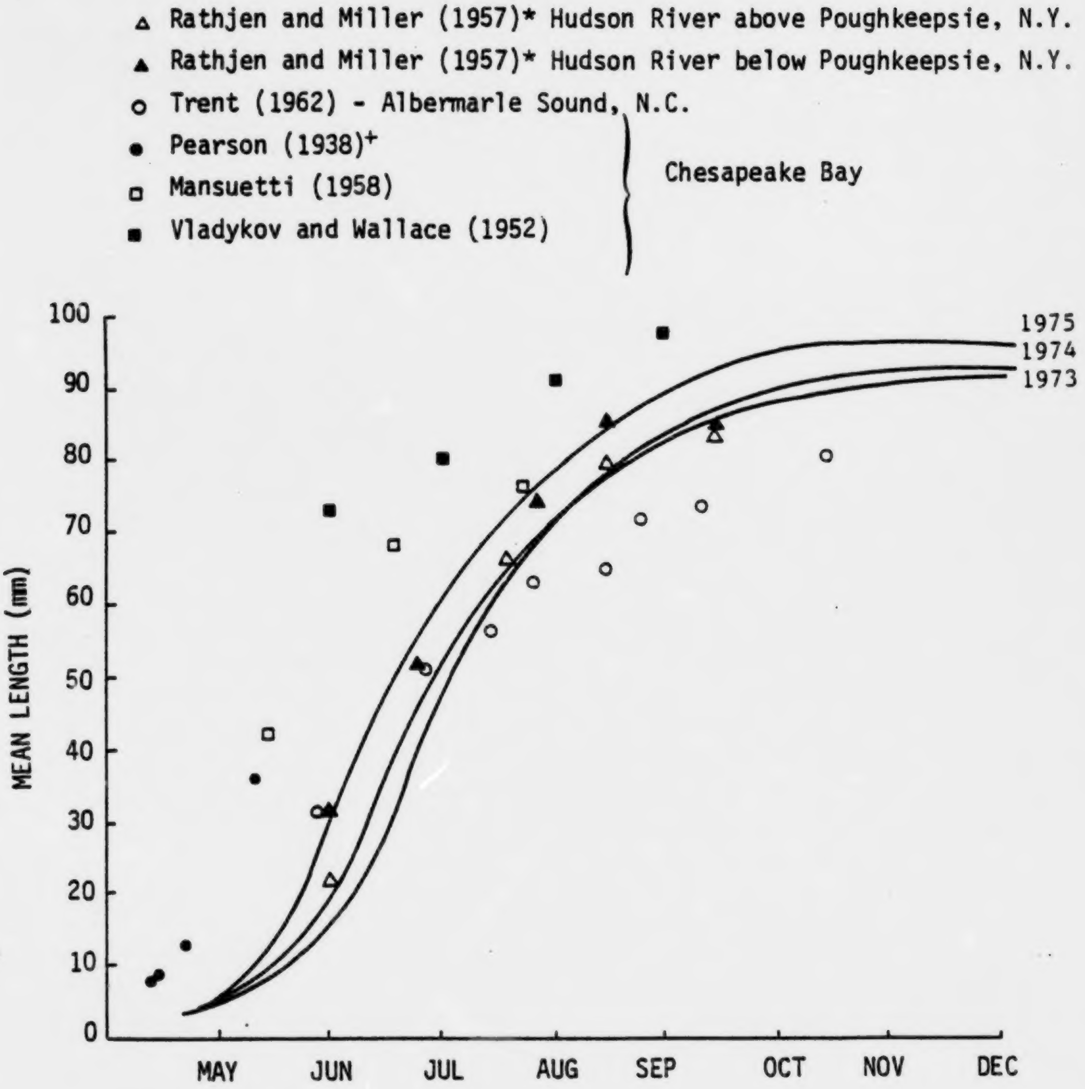
Normal Conditions and Physiological State

Normal growth for this stage has been presented for a variety of rearing conditions (See Figs. 25, 29, 30, 31, 38, and Table 21). Typical first year growth is shown in Figure 39 for bass taken from Hudson River, Chesapeake Bay and Albermarle Sound nursery areas. In Albermarle Sound, Trent (1962) observed that growth rate was almost linear among young-of-the-year striped bass from June to November. He calculated rates ranging from 0.272 to 0.433 mm/day for the five years of his study. In the Hudson River, Rathjen and Miller (1957) reported the greatest growth rate for young-of-year during June and July, which continued almost linearly to September-October. Texas Instruments (1976c) reported instantaneous growth rates (based on weight) calculated from young collected by beach seine during 1973, 1974, and 1975. The highest rates ranged from 0.0311 to 0.0407 for July-August, while the lowest ranged from 0.0145 to -0.0157 for October-November. The rate of growth in these locations was reported to be reduced during the winter months, increasing again in April and May during the spring warming of these river areas.

We calculated growth rates for various groups fed daily over broad temperature ranges during this study. Typically, juveniles exhibit a growth rate of 0.1 to 0.3 g per day at average seawater temperatures of 18-20°C, but only 0.001 to 0.002 g per day at 8-10°C. Yearlings grew about 0.2 g/day at 18-22°C, 0.5 g/day at 14-18°C, and only about 0.1 g/day at 8-12°C. Two year old growth was as high as 1.0 g/day at the intermediate temperatures and lower (0.5 to 0.3 g/day) for the extreme temperatures. We consider these to be indicative of healthy bass. It is evident that optimal growth occurs at temperatures of generally less than 18-20°C and greater than 10-12°C for bass during this stage.

Changes in the normal physiological state for bass of this stage from exposure to toxic substances have been shown by a number of investigators (see Section 12 for details). Sublethal exposure to benzene affects growth (Korn et al., 1976a), behavior (Korn et al., 1976b), and respiration rate (Brocksen and Bailey, 1973). Physiological responses from exposure to cadmium and mercury (Dawson et al., 1977) have also been reported.

Courtois (1974) reported physiological effects of temperature and salinity on yearling striped bass. Results showed saltwater acclimated bass had significantly lower blood characteristics, crude liver fat, serum K+, and higher serum Na+ levels than freshwater acclimated bass at a similar temperature. Increased temperature appeared to cause reverse effects in freshwater and saltwater acclimated bass. Freshwater acclimation at higher temperatures resulted in lower plasma protein and higher serum Na+ levels, while this resulted in seawater at lower temperatures. The metabolic and physiologic results are fully discussed by the author. In addition, he investigated sublethal effects of copper in relation to the established normal for bass in freshwater and seawater. This is discussed in Section 12.



* from Trent (1962)
+ from Mansuetti (1958)

Figure 39. Growth in length of young-of-the-year striped bass among 1973, 1974 and 1975 year classes in the Hudson River (solid lines) and mean lengths reported for other coastal populations (individual symbols). (McFadden, 1977a)

Effects of 0, 2, 4 and 10 mg/l suspended bentonite on oxygen consumption of juvenile striped bass at 18°C, 24 o/oo were reported by Peddicord et al. (1975). Regression coefficients at 0, 2 and 4 mg/l were not statistically different (3.628, 3.524, 3.653), however, the regression coefficient at 10 mg/l (5.361) was significantly different. These observations on the response of individual bass weighing about 7.3 g indicated a more rapid oxygen consumption rate at 10 mg/l suspended bentonite. The y-intercepts from regression analysis were -1.956, 0.580, -1.210, and -3.134 mg/g/hr for 0, 2, 4, and 10 mg/l concentrations, respectively. Uncontaminated suspended sediments taken from San Francisco Bay area showed no demonstratable effect at the concentrations tested (0-3.9 g/l at 25 o/oo, 12°C, and 8 ppm dissolved oxygen) on hematocrit levels of juvenile bass (Peddicord and McFarland, 1978). The amount of minerals in the stomachs of these bass showed slightly significant increases with increasing uncontaminated suspended sediment concentrations.

Dorfman and Westman (1970) observed the behavior of bass exposed to varying dissolved oxygen levels and temperatures. The bass preferred higher dissolved oxygen concentrations. Chittenden (1972) observed a maximum ventilation rate (gulps/sec) at 2-3 mg/l dissolved oxygen, declining at lower concentrations. The behavior pattern at low oxygen levels included restlessness at about 3 mg/l (16-19°C), followed by inactivity, loss of equilibrium and finally death at lower oxygen levels. Meldrim et al. (1974) observed avoidance temperatures of 22-26 and 31°C for juvenile bass acclimated at 17-18 and 26°C, respectively, and dissolved oxygen concentrations varying from high to low (4.0 mg/l) saturation levels. Bass acclimated at 18°C, 6.5 o/oo and pH of 7.2, generally avoided dissolved oxygen levels of 44-41% of saturation (4.0-3.8 mg/l).

Juvenile endurance and/or swimming behavior for various water velocities have been tested. Kerr (1953) reported that juveniles and yearlings endured 10 minute tests at 22°C swimming at velocities up to 30 cm/sec. This was equivalent to swimming at 6-12 body lengths per second. Texas Instruments (1975a) found that swimming-speed capabilities increased with increasing body length and water temperature. Critical swimming speeds for bass 143-224 mm TL ranged from 22.9-122.0 cm/sec at 18°C. Bibko et al. (1974) described effects of water velocity, air-bubble screens and intense illumination on the swimming behavior of juveniles at 11.1, 4.50 and 0.6°C. In still water bass swam randomly, but oriented and swam against oncoming current when exposed to water velocity. When given a choice in the range of velocities, bass swam against the current but always chose a path representative of the least water current. At 11 and 4.5°C, bass did not actively cross the air-bubble screen, but did drift passively through the screen at 0.6°C. The air-bubble screen reportedly deterred passage as effectively at night as during the day. These authors observed that intense illumination was only a temporary, or passive, deterrent to bass passage. Freadman (1978) showed that subadult striped bass use cyclic ventilatory movements at rest and at slow swimming speeds, but switch to ram gill ventilation at intermediate and high velocities. He determined the transition velocity was 45.6 cm/sec, or 1.36 to 3.14 body lengths per second (largest to smallest bass tested), at 15°C. Bass reserve white muscle for swimming at velocities above maximum sustainable speeds, and use red muscle for slow to cruising speeds. He concluded that they are strong swimmers.

Parasite infections and diseases recorded for striped bass are presented in Table 24. Most are reports for bass taken from natural environments or from freshwater rearing ponds. Bonn et al. (1976) summarize the common parasites and diseases of bass from pond culture systems providing some items of diagnosis and treatment under pond or raceway conditions. The most common treatment they suggest is Furacin as a bath (5-8 hours) at 22 ppm or active ingredient. This appears to be effective for bacteria, protozoans and Saprolegnia. It is recommended for use in ponds, tanks or in fish trucks. They note that it should not be used in galvanized containers and that it has been banned by the U.S. Federal Drug Administration.

Culturing our bass in ambient seawater reduced infection by many of the freshwater and brackish water parasites that have been reported (Table 24). However, we experienced infections of marine Trichodina sp. in addition to two other protozoan parasites not previously reported for striped bass. These are Cryptocaron sp. and Myxosporidin sp. Unlike the freshwater culturists, we relied on either malachite green or formalin for treatment of infected bass in our systems. Two useful references that review the effectiveness, usage, and efficacy of these two therapeutics are Nelson (1974) and Schnick (1973). Table 26 summarizes the treatment we recommend for the various parasite groups reported to infect bass. It is suggested that the baths recommended for fungus, protozoans, and crustacea be repeated every 2 or 3 days for a week to knock out these parasites completely.

The relationship between disease and environmental stress has recently been reviewed extensively by Sinderman (1979). Culture systems are potentially stressful environments. This is a situation the culturist must strive to avoid. Assessing the health of cultured fish should consist of routine examinations, especially visual observations, by the culturist. Hematological techniques (Blaxhall, 1972; Hickey, 1976; Wedemeyer and Yasutake, 1977) and other clinical methods have been proposed as tools for this assessment. (Wedemeyer and Yasutake (1977) provide a very good description of methods and their interpretation.) A number of studies have reported some of the hematological values for wild and laboratory reared striped bass (see Section 5) to which comparisons of values from cultured bass may be made. In some cases only histopathological examinations and/or simple staining of impressions or smears will allow disease or parasite diagnosis. Ribelin and Migaki (1975) provide additional descriptions to those given below of both infectious and noninfectious diseases.

During the course of this study, we submitted fish or posted tissue from both bass in our holding tanks and those collected in the field (juveniles and adults) to the URI Marine Pathology Laboratory. At the Laboratory the material was examined, generally histopathologically, and a diagnosis or findings were reported to us. Table 27 presents a summary of the findings by 'agent' responsible for the lesions observed histopathologically from samples submitted.

Lesions seen in the gill accounted for the majority of the pathological

TABLE 26. TREATMENT RECOMMENDED FOR SOME OF THE PARASITE GROUPS COMMON TO STRIPED BASS (SEE TABLE 24. FOR SPECIFIC PARASITES)

Type of Parasite	General Appearance of Infection	Treatment Recommended
Virus and Bedsonia	white, wart-like growths on skin and/or gills	none known
Bacteria	varies with specific organism; diagnosis requires histopathological preparation of tissues	varies with organism of infection; antibiotics administered in water or food
Fungus	white cottony growth on skin and fins	bath, malachite green at 1-3 mg/l for hour
Protozoans	varies with organism, but some are visible at high power from scrapings and staining of gills and/or skin	bath, formalin at 1:4000 (250 ppm) for 30 min. (to 60 min.); or malachite green 1:200,000-400,000 for 30 min.
Trematodes	organism visible under low power magnification in gills or gut	salt bath (at least 1%) if in freshwater; formalin bath at 1:4000 for an hour
Cestodes and Nematodes	internally visible if fish is sacrificed	usually low key and not a problem unless fish is under stress
Crustacea	whiteish spots visible at low power on gills and/or skin	bath, formalin at 1:4000 for 30-60 min.; salt bath if in freshwater
Hirudinea	visible attached to skin or fins	low key; salt bath
Mollusca	visible (valves at low magnification) on gills	salt bath

TABLE 27. INCIDENCE OF LESION TYPE

Etiologic Agent	Number of Cases (n=150)
Epitheliocystis	44
Ergasilus labracis	35
Philometra rubra	12
Microcotle macrura	1
Trematodes (unclassified)	12
Diplostomulum sp. (metecercaria)	10
Pomphorhynchus rocci	23
Stephanostomum tenue	5
Lymphocystis	3
Saprolegnia sp.	1
Aeromonas sp.	2
Mycobacterium sp.	2
Fibropapilloma	1
Nephroblastoma	1
Trichodina davisi	25
Trichodinella sp.	12
Glossatella sp.	1
Scyphidia sp.	2
Oodinium sp.	5
Cryptocaryon sp.	2
Myxosoma morone	4
Myxosporidiosis (unclassified)	9
Glochidia	5

findings from material examined. Lesions were present in 102 of the 150 gills submitted. These represented inflammatory changes (necrosis and cellular infiltration by mononuclear and neutrophilic cells), circulatory changes (hemorrhage, edema and congestion), growth disturbances (hypertrophy, hyperplasia and squamous metaplasia), and physical lesions (cyst formation). Figure 40 presents observations of normal gill filaments as well as representative lesion responses among striped bass.

A wide variety of lesions could be noted involving the skin and at times, the underlying skeletal muscle. Acute and chronic dermatitis was associated with dermal congestion, hemorrhage,ulceration and necrosis. The origin of the lesions seen involving the skin included bacteria (Aeromonas and Mycobacteria), virus (lymphocystis disease), parasites (Figure 41 a,b, and c: Trichodiniasis, Scyphidiasis, Cryptocaronasis), physical (cysts of metacercarial origin) and fungal (Saprolegnia sp.). One case involved a neoplasm of the skin (fibropapilloma, Fig. 42 c). Fifteen cases had abnormalities relating to diseases of the skin and underlying skeletal muscle. Primary involvement of the skeletal musculature occurred in 5 cases. Inflammatory lesions (acute and chronic myositis), necrotic myositis, acute myolysis, and granulomatous myositis were the primary lesions. These could be attributed to a variety of etiologies including bacteria (Aeromonas liquefaciens, Mycobacteria), parasites (metacercarial cysts and granulomas) and fungi (Saprolegnia sp.). Twelve cases had abnormalities of bone or cartilage. Seven cases were associated with lesions of cartilage of parasitic origin (Fig. 41 d and e: Myxasporidian cysts and Myxosoma morone).

Lesions associated with the nares (Fig.42a) were composed of inflammatory (chronic and acute rhinitis of mononuclear cell response), physical (cyst, Fig. 42 c), and parasitic (Trichodiniasis, Fig. 42 b) types. Four cases of 150 submitted involved the nares. Tissue responses of the nares were similar to those of the gill.

Abnormalities of the kidney were not common but striking when they appeared in histopathological sections. The majority of diagnoses of disease involving the kidney were related to parasitic infestations (protozoal cysts, granulomas, and parasitic cysts of nematodes, trematodes, and acanthacephala). Renal tubular dilation was observed to occur in two cases with proteinaceous casts present in the renal tubules. Neoplasia (nephroblastoma Fig. 42d) occurred in one case involving a female striped bass (94.5 cm FL) taken on the spawning grounds. This is a rare neoplasm and the second report of such in a striped bass (see Table 24). Abnormalities involving the head kidney were seen in 8 cases. All cases involved parasitic cysts or granulomas in the parenchyma of the head kidney.

The spleen had disease changes associated with follicular hypertrophy, follicular atrophy, inflammation (granulomas of parasitic origin) and cyst formation (metcercarial cysts). The spleen was involved in 21 cases of abnormalities,the majority of which were parasitic in origin.

Many types of lesions were observed in the liver. These included inflammatory reactions (necrosis, fibrosis, granulomas, chronic cellular

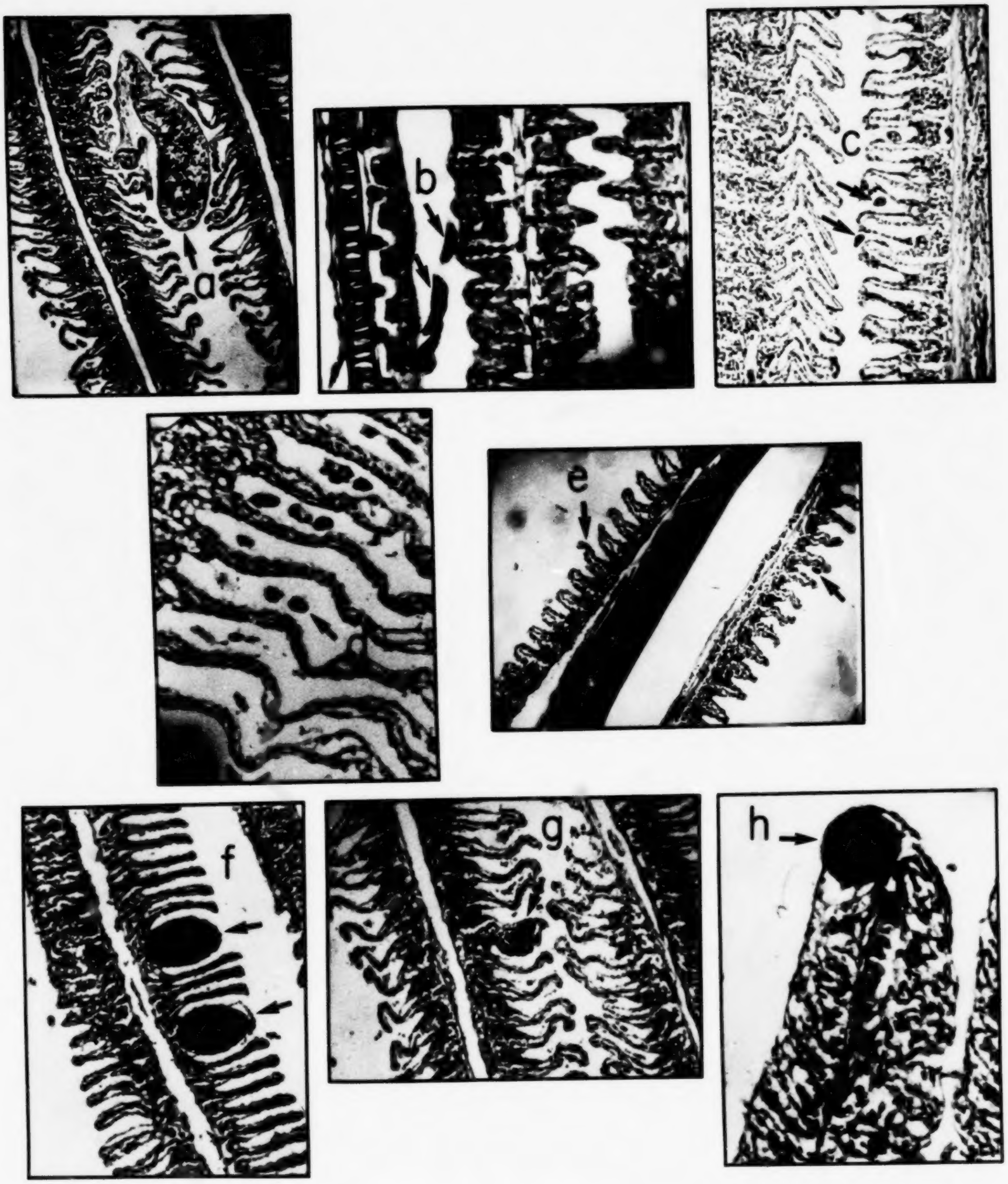

a
b
c
e
f
g
h

Figure 40. Composite illustrating lesions and infections of the gill filaments of striped bass observed from histopathologic examination: a. trematode and sections of normal gill filaments; b. *Trichodina* and hypertrophied gill filaments; c. *Oodinium* infection; d. *Glochidia* infection; e. *Glossatella* sp. infection; f. early epitheliocystis; g. aneuresm; and h. myxosporidiosis cyst.

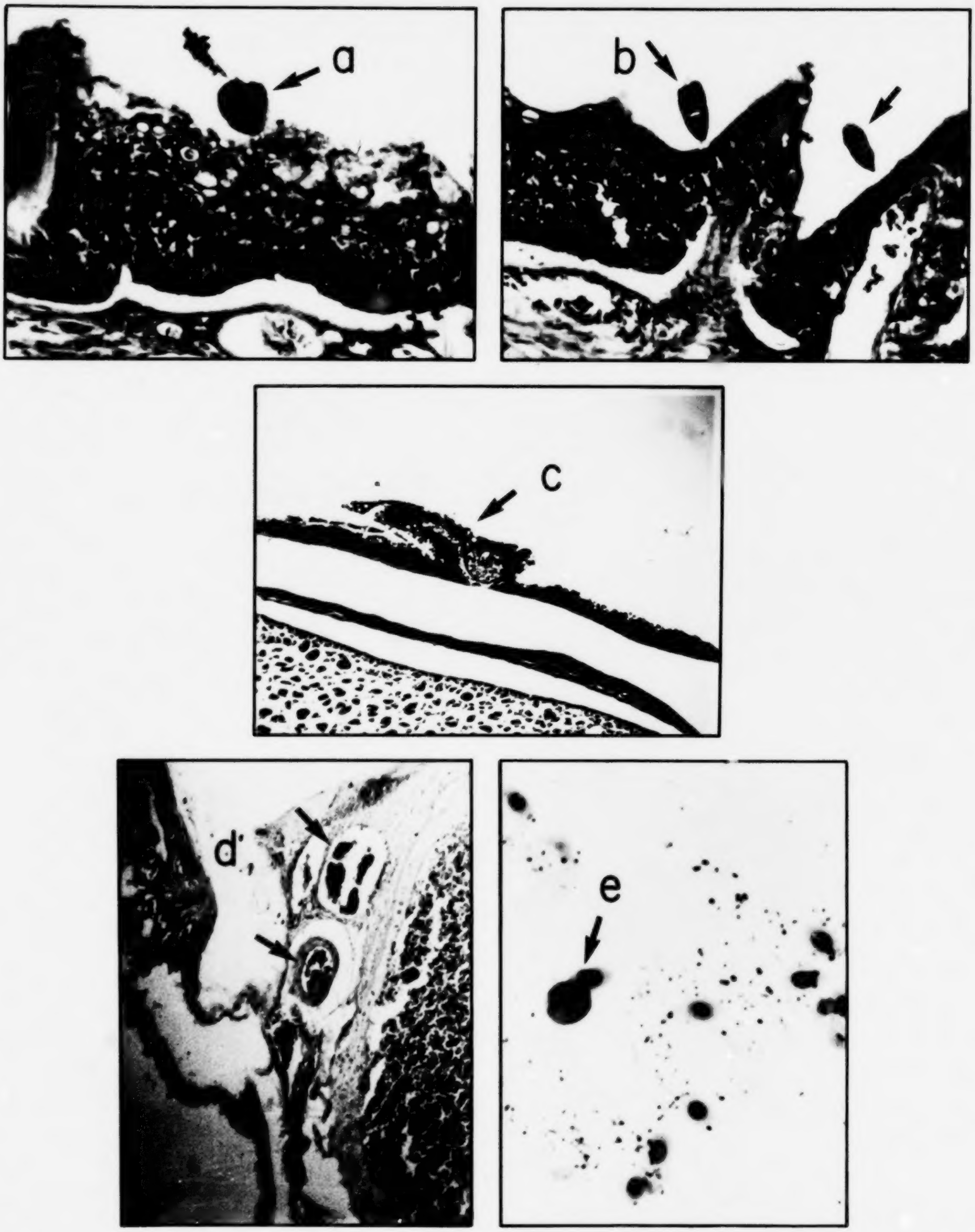
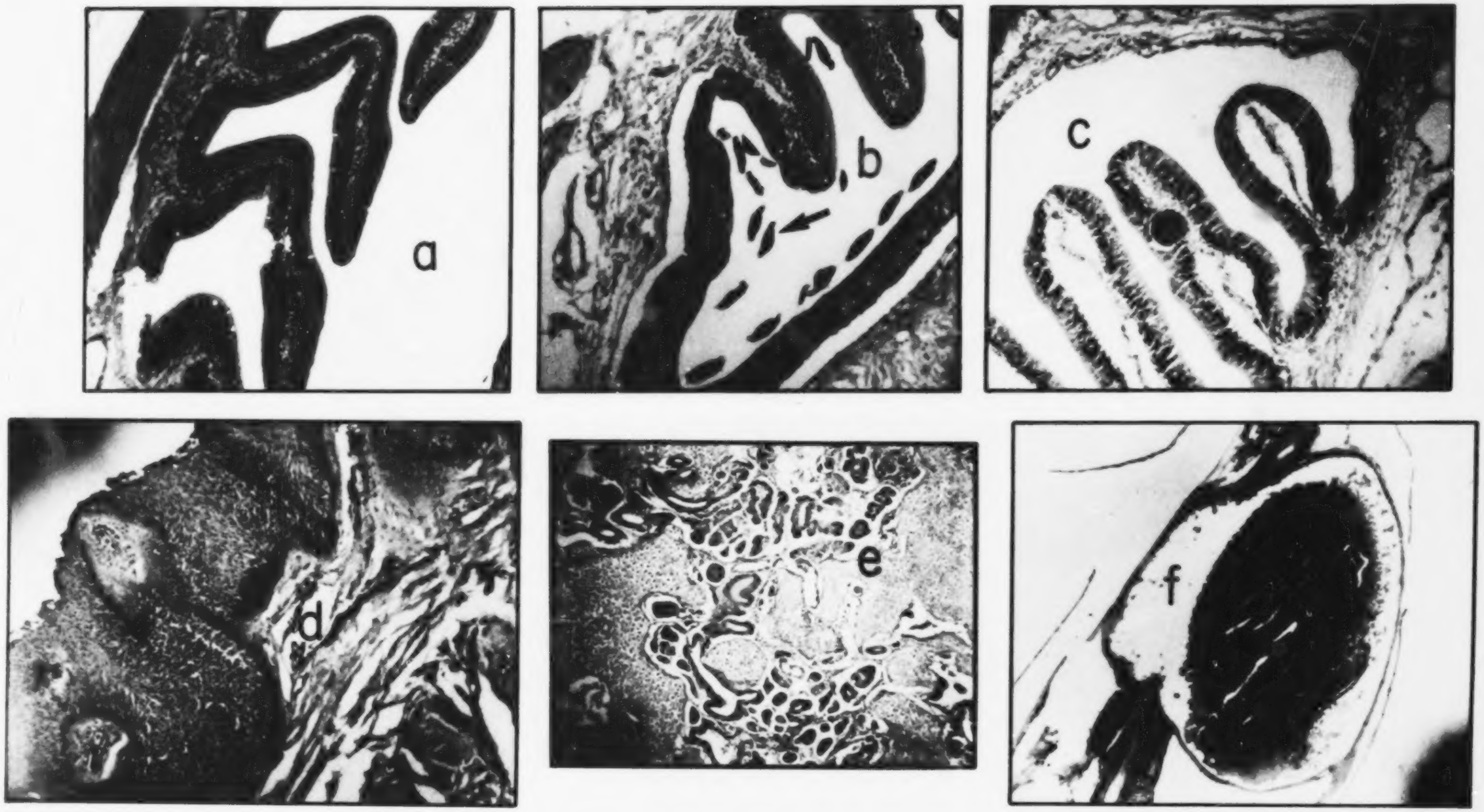

a
b
c
d,
e

Figure 41. Composite illustrating some parasites of striped bass skin and cartilage: a. *Trichodina* sp.; b. *Scyphidia* sp.; and c. *Cryptocaryon* sp. infecting skin areas; d. myxosporidiosis cyst in cartilage; and e. *Myxosoma* sp. stained with Giemsa.


a
b
c
d
e
f

infiltration of mononuclear-phagocytic cell origin), metabolic changes (fatty infiltration and hyperplasia) and physical abnormalities (parasitic cysts). Twenty cases involved disorders of the liver.

A congenital developmental cyst of the yolk sac was recorded in a 4 day old bass larva (Fig. 42 e).

Figure 42. Composite illustrating lesions of the nares (a-c) and other neoplasma (d,e) and cysts (f): a. normal nares; b. Trichodina sp.; and c. epitheliocystis in nares; d. fibropapilloma of the mandible; e. nephroblastoma from dorsal wall of swim bladder; and f. cyst in yolk sac of larva hatched in the laboratory.

SECTION 11

RECOMMENDED CULTURE METHODS AND BIONOMICS: ADULT

DESCRIPTION OF STAGE

This stage encompasses that portion of the striped bass life history from sexual maturity to death. The primary occurrence during this stage is spawning.

This species is heterosexual, although hermaphrodism has been reported. Schultz (1931) reported a 5.44 kg, 60 cm individual from Oregon waters apparently with both maturing ovary and testis. Westin (1978) reported one hermaphrodite from Rhode Island waters. This was a 52 cm and 1.63 kg immature individual. Sexual dimorphism has not been observed for this species.

Age at maturity for males and females captured from different areas is summarized in Table 28 . Males are mature by their third year or at a length of about 30 cm FL and a weight of about 400-500 g. Most females mature during their fifth, sixth or seventh year or at a length of approximately 50 cm FL and a weight of 1-2 kg (Merriman, 1941; McFadden, 1977a; Scofield, 1931). Some, however, mature during their third or fourth year (Lewis, 1962). Usually the larger, or older, bass are female. The largest bass recorded weighed 56.3 kg and was taken commercially at Edenton, North Carolina (Raney, 1952). The largest bass reported taken by rod and reel was 33 kg (Moss, 1974).

Length-weight relationships reported from different areas are summarized in Table 29. Throughout their range it appears that after bass mature, the males of a given length weigh less than females of the same length (Merriman, 1941; Mansueti, 1961). Growth is more rapid during the second and third years of life, or before maturity (see Section 10) than in later years. Growth in length of both sexes is summarized by age groups in Table 30 for a number of areas. Bass over 14 or 15 years are a rarity.

Most of the description of gonad development during this stage has been reported for females. Table 31 summarizes the relationship between gonad size and fecundity in females, to body weight, length and age for both sexes of bass captured in different regions. Most of these data were taken during the spawning season. Hollis (1967) determined weights of both right and left ovaries among 28 bass. His data shows that in 14 of these pairs the left ovary weighed less than the right one. Vladykov and Wallace (1952) reported the ratio of body weight to gonad weight for males and females in Chesapeake Bay. Texas Instruments (1973) reported this

TABLE 28. AGE AND SIZE AT FIRST MATURITY FOR STRIPED BASS

Area	Age (years)[a]		Length (mmFL)		Weight (kg)		Author
	males	females	males	females	males	females	
					(based on L-wt eq.)		
Southern New England	3	5	(350)[b]	570	—	—	Merriman (1941)
Hudson River	-	5	—	551	—	—	Texas Instruments (1975b)
Delaware River	3	4	303	—	0.3	—	Bason (1971)
Upper Chesapeake Bay	3	4	330	515	—	—	Pearson (1938)
Potomac River	2	4	330	450	0.6	1.5	Wilson et al. (1976)
Lakes Marion and Moultrie	-	4	610	—	—	—	Scruggs (1957)
Sacramento-San Joaquin Rivers	-	5	—	535	—	1.8	Scofield (1931)

[a] Age at which at least 50% are mature.

[b] Number in parenthesis is best approximation available from data given by the author.

TABLE 29. LENGTH-WEIGHT RELATIONSHIP FOR STRIPED BASS

(Log_{10} weight = a $\log_{10}$ length + b)

Area of Capture	Number in Sample	Sex[c]	Units[d]	Slope (a)	Intercept (b)	Years of Samples	Source
Maine rivers	216		1	3.049	-3.420	1964-1965	Davis (1966)
Massachusetts	400		1	2.9616	-3.2838	1956-1959	Frisbie (1967)
Rhode Island	475		7	2.851	-4.644	1973-1975	Authors' data
New York, Hudson River	2678		8	2.839	-1.825	1971-1972	Lawler *et al*. (1974b)
	51	M	5	2.956	-4.880	1972	Texas Instruments (1973)
	31	F		3.130	-5.340		
	1120	Y		2.940	-4.886		
	100	M	5	3.265	-5.750	1974	Texas Instruments (1975b)
	83	F		3.424	-6.180		
	10	Y		1.207	-3.329		
Delaware River	100	M	4	3.000	-4.950	1969	Bason (1971)
	100	F		2.911	-4.736		
Chesapeake-Delaware Canal	117		4	3.0501	-5.0001	1974	Bason *et al*. (1975)
Chesapeake Bay	207	M	2	3.234	-2.406	1957-1958	Mansueti (1961)
	315	F		3.153	-2.238		
Nanticoke River, Md.	89		7	2.894	-4.665	1974-1975	Authors' data
Potomac River, Md.	1034	M&F	4	2.9381 (given in ln)	-10.6953	1975	Wilson *et al*. (1976)
Rappahannock River, Va.	1364	Y	4	3.073	-5.081	1969-1971	Kerby (1972)
Albemarle Sound, N.C.	3097	Y	6	2.9198	-1.8462	1955-1961	Trent (1962)
Keystone Reservoir, Okla.	148		2	2.7381	-2.9875	1969-1971	Erickson *et al*. (1972)
California	1089		1	3.0038	-2.1393	1957-1958	Robinson (1960)
Coos Bay, Ore.	1329		3	2.90679	-4.588	1949-1950	Morgan & Gerlach (1950)

[c] Sexes combined unless stated otherwise, where M-male, F=female, Y=young-of-year & yearling.

[d] Units of original measurements for weight and length terms:
1=pounds, inches FL; 2=pounds, inches TL; 3=pounds, cm FL; 4=gm, mm FL; 5=gm, mm TL; 6=mg, mm TL; 7=Kg, cm FL; 8=gm, cm.

TABLE 30. COMPARISON OF GROWTH (mm) OF STRIPED BASS FROM VARIOUS AREAS[a]

Area of Capture		Number in Sample	AGE GROUP																	Author
			I	II	III	IV	V	VI	VII	VIII	IX	X	XI	XII	XIII	XIV	XV	XVI	XVII	
Maine, MCFL		216	150	297	408	488	556	617	658	—	—	—	—	—	—	—	—	—	—	Davis (1966)
Massachusetts, MCFL		1056	150	305	437	551	648	737	810	876	927	970	1006	1036	1067	1087	1105	1128	1158	Frisbie (1967)
Rhode Island, MMFL		238	—	—	425	503	576	648	720	863	893	954	997	1001	—	1125	—	—	—	Authors' data
Connecticut, MCFL-4; MMFL-9		25	125	235	365	450	530	610	685	750	820	—	—	—	—	—	—	—	—	Merriman (1941)
Hudson River, N.Y., MCFL[b]	M	98	103	228	339	434	517	592	645	681	666	—	—	—	—	—	—	—	—	Texas Instruments (1974a)
	F	67	104	232	358	459	552	637	708	770	815	855	909	799	846	—	—	—	—	
Delaware Estuary, MCFL		112	102	218	319	430	530	630	708	795	869	919	992	1045	—	—	—	—	—	Bason (1971)
Chesapeake Bay, MCFL	M	224	135	297	381	422	500	595	704	754	831	876	906	—	—	—	—	—	—	Mansueti (1961)
	F[c]	520	124	292	380	467	556	645	724	782	856	899	935	1006	983	1044	—	—	—	
Potomac River, Md., MMFL	M	843	—	331	381	431	508	654	766	784	864	839	—	—	—	—	—	—	—	Wilson, et al. (1976)
	F	218	—	338	389	449	549	696	785	829	884	926	936	960	1030	1093	—	—	—	
Nanticoke River, Md., MMFL		75	—	—	376	450	553	579	736	868	865	945	1007	—	—	—	—	—	—	Authors' data
Roanoke River, Va., MCFL[d]		101	132	307	475	587	658	693	724	747	780	800	—	—	—	—	—	—	—	Domrose (1963)
Roanoke River, N.C., MFL	M	2403	—	356	424	465	503	551	594	632	813	—	—	—	—	—	—	—	—	Trent and Hassler (1968)
	F	683	—	—	465	513	544	602	650	671	724	742	762	965	902	—	—	—	—	
North Carolina, offshore, MCFL		277	137	267	392	491	582	667	750	815	868	913	953	998	1054	1098	—	—	—	Holland and Yelverton (1973)
Santee-Cooper Reservoir, S.C. MCFL		322	216	399	503	582	655	724	767	—	—	—	—	—	—	—	—	—	—	Stevens (1958)
MCFL[d]		414	170	356	465	528	599	655	719	772	826	—	—	—	—	—	—	—	—	Scruggs (1957)
Savannah River, Ga., MCFL	M	59	147	260	363	431	526	635	695	746	671[e]	711	754	793	823	—	—	—	—	Smith (1970)
	F	213	152	288	386	481	592	688	787	856	914	953	991	—	—	—	—	—	—	
Sacramento-San Joaquin, Calif. MCFL	M	204	106	251	371	445	516	563	612	—	—	—	—	—	—	—	—	—	—	Scofield (1931)
	F	269	106	247	370	460	542	612	680	—	—	—	—	—	—	—	—	—	—	
MMFL	M	314	98	286	373	463	490	541	610	685	805	785	—	—	—	—	—	—	—	
	F	972	97	264	346	458	535	605	686	777	795	870	947	900	1030	1080	—	—	—	
MCFL	M	385	104	249	386	493	566	622	671	726	762	—	—	—	—	—	—	—	—	Robinson (1960)
	F	295	104	249	389	500	594	685	747	800	836	—	—	—	—	—	—	—	—	
Coos River, Ore., MMFL		NA	—	—	368	483	575	635	695	730	760	—	—	—	—	—	—	—	—	Morgan and Gerlach (1950)

[a] MCFL = mean calculated fork length; MMFL = mean measured fork length, MFL = mean fork length; sexes combined unless indicated otherwise.

[b] Converted to fork length (FL = 4.6 + 0.902 TL).

[c] Contains females and bass of unknown sex.

[d] Converted from MCTL by factor of 0.93 (Mansueti, 1961).

[e] This negative growth figure is the result of a single, unusually small specimen for its age being used for the calculation (Smith, 1970).

TABLE 31. RELATIONSHIP OF GONAD WEIGHT, EGG NUMBER, BODY LENGTH AND BODY WEIGHT AMONG STRIPED BASS OF VARIOUS AGES CAPTURED IN A NUMBER OF AREAS

Area and Age	Number in Sample	Gonad Weight total (gm)	Number of Mature Eggs (mean or range)	Body Weight (kg)	Body Length (FL,cm)	Author
Hudson River, New York						
6	2	—	451,000	—	55.1	Texas Instruments (1973)
8	14	—	1,548,000	—	80.9	
10	4	—	1,841,000	—	88.9	
Upper Chesapeake Bay, Maryland						
—	1	—	1,337,000	5.897	70	Pearson (1938)
Chesapeake Bay, Maryland						
4	10	58.5	68,239	1.996	51.2	Jackson and Tiller (1952)
6	7	594.0	856,257	5.897	71.26	
8	13	988.0	1,682,292	7.212	85.0	
10	2	1319.0	2,510,349	9.752	92.1	
males						
(2½)*	16	27.1	—	0.55	34.0	Vladykov and Wallace (1952)
(3-4)	11	89.9	—	1.461	44.2	
females						
(5½)	4	120.0	—	2.50	56.5	
(8½)	3	755.0	—	8.67	83.2	
Nanticoke River, Maryland						
4	3	71.6-154.2	201,000-555,000	1.13-2.09	42.7-53.6	Hollis (1967)
5	3	245.0-336.6	601,000-857,000	3.45-4.63	65.3-69.3	
10	1	1650	2,207,000	14.97	100.3	
Transquaking River, Maryland						
4	3	118-149.7	252,000-416,000	1.81-1.86	49.8-52.3	Hollis (1967)
6	2	427.7-543.9	898,000-1,319,000	4.63-4.76	69.6-71.1	
Elk River, Maryland						
5	2	275.3-347.5	494,000-591,000	3.13-3.31	60.5-61.5	Hollis (1967)
10	1	1897	2,310,000	11.57	90.9	
12-15	7	1966-3476	2,248,000-4,136,000	11.88-25.85	91.9-119.4	
Potomac River, Maryland						
12-14	4	2142-3011	3,257,000-4,864,000	20.14-25.63	108.7-115.6	Hollis (1967)
Roanoke River, Weldon						
—	1	—	14,000	1.361	—	Worth (1904)
—	1	—	265,000	2.041	—	Merriman (1941)
—	1	—	3,220,000	22.68	—	Worth (1904)
Roanoke River, North Carolina						
4	13	—	320,000	1.81-2.22	50.8-53.1	Lewis and Bonner (1966)
6	8	—	454,000	2.73-3.13	55.9-58.2	
10	2	—	1,090,000	6.35-6.76	71.1-73.4	
Offshore, North Carolina						
8	4	126-867	1,044,230-2,221,821	7.3-8.8	80-84	Holland and Yelverton (1973)
9	13	67-1253	1,067,472-3,715,939	7.7-13.6	82-98.1	
10	4	180-2123	1,995,974-4,057,059	9.0-19.0	89.2-109	
12	2	663-914	3,304,497-3,511,038	12.2-12.7	95.0-98.7	
Coos Bay, Oregon						
—	1	—	900,000	3.99	—	Morgan and Gerlach (1950)
—	1	—	4,775,000	22.68	—	

*Numbers in parentheses are best estimates from data given by the author.

information for the Hudson River and Wilson et al. (1976) for the Potomac River. Their data are summarized by percent of gonad weight to body weight for each sex during development:

	Chesapeake Bay		Hudson River		Potomac River	
	M	F	M	F	M	F
immature	-	0.7	0.2-1.2	0.4-1.0	0.6-2.3	0.4-0.5
maturing	-	1.7	-	-	3.2-7.3	0.7-5.6
prespawning	5.0	4.8	1.4-11.1	1.1-16.7	1.4-13.6	6.7-10.3
spawning	6.3	8.3			4.1-9.1	11-16
spent	-	1.3	-	-	0.7-2.0	3.6-4.8

The number of eggs produced by the females (i.e., fecundity) is highly correlated with weight, length and age. The number of eggs increases with age, although there is considerable variability between individuals of the same age group. An immature ovary contains small ova 0.04 to 0.23 mm in diameter. A mature ovary contains both small and large ova. The large ova average 0.16 to 0.76 mm in diameter, increasing to 1.0 to 1.35 mm at spawning (Chadwick, 1965; Jackson and Tiller, 1952; Lewis, 1962; Merriman, 1941). As they mature, the ova and ovaries change in color from cream to orange to pale, or grass green. Fecundity data, mainly from individual females, are plotted in Figure 43.

There appears to be some suggestion of abnormal egg development among hormone induced spawnings. The success of hatching appears better among naturally spawned eggs than among artifically spawned eggs. An indication of this is shown in Table 32 from our studies and those of Shannon (1970). Although the use of hormone injections for artificial spawning affects the successful development of embryos to hatch normally, those embryos that hatch appear, under proper conditions, to grow into normal larvae. Until the exact nature of the apparent hormone effect on embryo development is ascertained, natural development and spawning relying on control of abiotic and biotic factors in the maturing bass environment outlined below is recommended.

Vincent et al. (1969) reported ovarian ribosomal cistion amplification in striped bass.

Clarke (1973) detected sodium retaining capacity of prolactin in the pituitary of striped bass.

NATURAL HABITAT

Adult striped bass are found along the Atlantic, Gulf and Pacific coasts of North America. They are important to sport and commercial fishermen within their range. For example, the estimated marine recreational catch for 1970 was 38.04 metric tons (NMFS, 1976). The 1974 total commercial landings for the United States were given as 5089.8 metric tons (NMFS, 1976) and 5097 metric tons (FAO, 1975). Many states have size limits

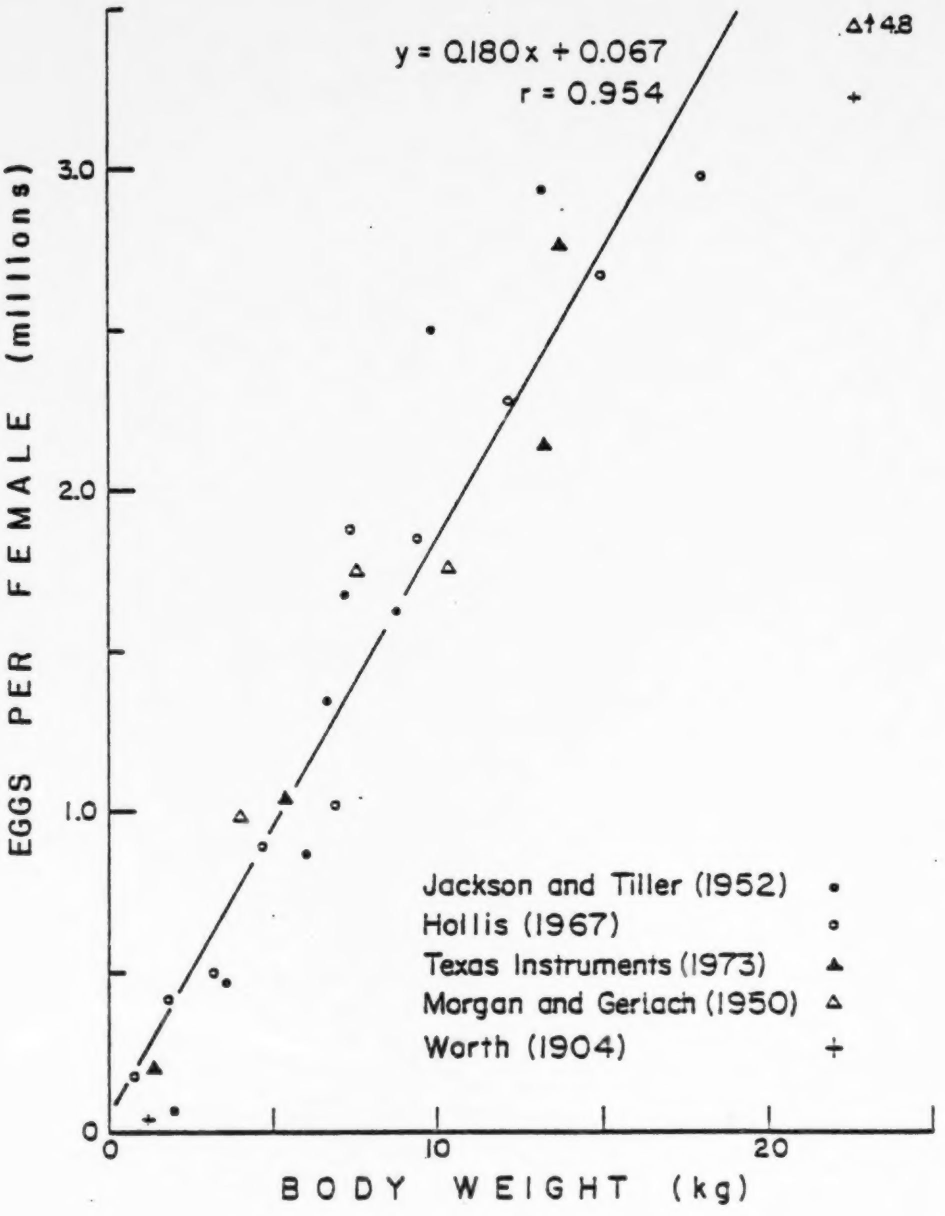


Figure 43. Fecundity of the striped bass in relation to individual weights. The regression line indicated in the figure was fitted by the authors to the data shown. Similar regression equations are given in Section 13.

TABLE 32. PERCENT SURVIVAL THROUGH HATCHING OF STRIPED BASS EGGS FROM ARTIFICIAL AND NATURAL SPAWNINGS

Incubation Salinity[*] (o/oo)	Incubation Temperature (°C)			
	16	18	20	21
Artificially induced spawning				
0	58.5 (561)[+]	64.3 (280)	-	4.7 (536)
5	1.2 (249)	-	7.4 (244)	5.4 (185)
10	19.3 (165)	-	11.6 (215)	11.6 (205)
15	31.2 (160)	-	0	0
Naturally matured spawning				
0	77.9 (384)	71.0 (473)	-	71.5 (421)
5	90 (10)	-	90 (10)	-
10	90 (10)	-	90 (10)	-
15	90 (10)	-	80 (10)	-

[*] Percent survival at 0 o/oo and 16 (60°F), 18 (65°F), and 21°C (70°F) reported by Shannon (1970). Survivals at the other salinity-temperature combinations are results of this study.

[+] ( ) = number of eggs per treatment.

governing bass fishing which vary in minimum from 25 to 46 cm FL (see Section 14).

Adults migrate primarily along the Atlantic and Pacific coasts for spawning and feeding. It appears from tagging studies that the striped bass from Chesapeake Bay and north tend to remain associated with a spawning river. Also, the tendency for bass less than 2-3 years of age not to undertake long coastal migrations seems to be supported. Coastal migrators appear in general to be post-spawning striped bass from the lower Chesapeake tributaries, Roanoke River and Albemarle Sound, supplemented in the Middle Atlantic and southern New England waters by Delaware and Hudson River bass. All indications are that striped bass from Albemarle Sound do not participate in the long coastal migrations of the bass from northern waters, although those off Cape Hatteras, North Carolina, may participate. Bass from South Carolina, Georgia, and Florida waters, as well as those from the Gulf coast appear to have foregone coastal migrations in favor of the fresh and brackish waters of their "home" rivers. The Pacific coast striped bass migrate extensively, but generally within San Francisco Bay and its tributaries. Coastal migrations of the nature seen on the Atlantic coast are not evidenced from tag returns along the Pacific coast (see Section 6 for details).

The striped bass is anadromous, spawning once a year generally from April to June throughout its spawning range. Ripe females, within hours of spawning, have, however, been observed off the coast of New England during late June and July. Data concerning spawning seasons, including temperature at spawning times, are given earlier in Table 8. Other spawning sites include the St. Lawrence River (Leim and Scott, 1966), rivers in Nova Scotia and New Brunswick (Bigelow and Schroeder, 1953), Chester, Choptank, Blackwater, Transquaking, Wicomico, Pocomoke and Patuxent Rivers in Maryland (Hollis, 1967), St. Johns and Appalachicola Rivers in Florida (Barkuloo, 1970), lower Colorado River, Arizona-California, Nevada (Edwards, 1974) and Coos River in Oregon (Morgan and Gerlach, 1950).

Males are first to arrive on the spawning grounds in early spring. Vladykov and Wallace (1952) reported up to 83% males in the Choptank River, Maryland, in March of 1937. However, for the year 1936-1937, they reported 55% of the 1211 bass they sampled from commercial fishermen in Chesapeake Bay rivers were male. The females appear to spend more of their time offshore, for Holland and Yelverton (1973) found only 11.8% males in 1970-1971 off the North Carolina coast. Most of the females taken on their trawl samples were almost ripe. Trent and Hassler (1968) reported sex ratio estimates for the spring of 1963, 1964, and 1965 at 69.7%, 85.1%, and 76.9% males, respectively, among gill netted striped bass in the Roanoke River.

After the females arrive on the spawning grounds, numerous "rock fights", as matings are known, can be observed. A single spawning female, swimming near and breaking the surface, surrounded by 10 to 50 (Merriman, 1941) or 5 to 20 (our observations) spawning males account for this behavior. This activity occurs anytime during the day, but is most often seen at dawn or dusk. The ripe eggs are released into the water where they are externally fertilized.

ENVIRONMENTAL REQUIREMENTS

The major abiotic and biotic factors important to the maintenance of adults are outlined in Table 33. Requirements associated with spawning are discussed at the end of the biotic factors section.

Abiotic Factors

Tagatz (1961) found adult striped bass acclimated to temperatures within the range of 6.7 to 30°C were tolerant to abrupt changes from salt to freshwater at differences in temperature over the range of 7.2 to 26.7°C. Four year old striped bass acclimated at 20°C avoided temperatures of 26.7°C and had a 48 hour LD50 of 31.5°C (Gift and Westman, 1971). We observed the general hardiness of adults during our maintenance of continuous laboratory populations throughout four years with little difficulty. The groups were generally in ambient seawater (0 to 26°C), although changes to freshwater during winter months have occurred within a day with no losses. The adults we have lost during the winter succumb to sea water temperatures of -1.0°C or less for periods longer than a few hours. Adults can survive 0°C sea water (28-30 o/oo) temperatures for a few days, although it is probably physiologically stressful. The warmest temperatures we experienced in our ambient sea water system were 26-28°C during early August (see Figure 3). While no mortalities occurred, the general behavior of the adults was not really normal. They swam alternately fast and slow, and did not feed steadily as they did at slightly lower temperatures of 20-24°C. It thus appears that seawater temperatures of over 26°C are probably stressful. The broad tolerance to salinities from fresh to sea water is evidenced by their anadromous spawning and coastal feeding migrations. The temperature and salinity requirements during spawning were discussed in the embryo section and below.

Adults require high dissolved oxygen concentrations especially during warmer periods due to increased respiration. Body weight markedly affects respiration (see Figure 44) among adult bass. Thus, at warmer temperatures when actively feeding, groups of adult bass can quickly deplete dissolved oxygen concentrations unless efforts are made to maintain them continually at saturation levels.

Biotic Factors

Food consumption is, together with density, one of the most important factors in the environment of any cultured species. The amount consumed must satisfy the needs of maintenance, growth and activity. The food presented for consumption must, of course, be palatable in order to be acceptable as a diet. Food items found to be preferred among adults were indicated in Table 20. Table 34A shows a range of daily satiation levels for adults during our feeding studies. The foods were fresh frozen, and cut into pieces of acceptable size to the bass. The menhaden was fed less heads. As the temperatures dropped, so did food consumption. At 4-5°C, adult feeding essentially ceased. However, for temperatures of 10 to 22°C, a feeding frequency of 3 to 5% of live weight daily (Table 33) is suggested. Based on the estimated evacuation time for a meal at these temperatures

TABLE 33. ENVIRONMENTAL REQUIREMENTS OF STRIPED BASS ADULTS

	ABIOTIC FACTORS	
	Survival Range	Optimum Conditions
Temperature	0-26°C	>10 & <24°C
Salinity	0-30 o/oo	10-30 o/oo
Dissolved oxygen	>8% (6.0 mg/1 @ 18°C)	air saturated
Light	no adverse effect	natural photoperiod
Turbidity	insufficient data	insufficient data
	BIOTIC FACTORS	
Diet	3-5% body weight (wet) daily	
Density	2.4 g/1 maximum (110 kg/45 m^3)	
Predators	none except man	
Disease and Parasites	for summary see Table 24	

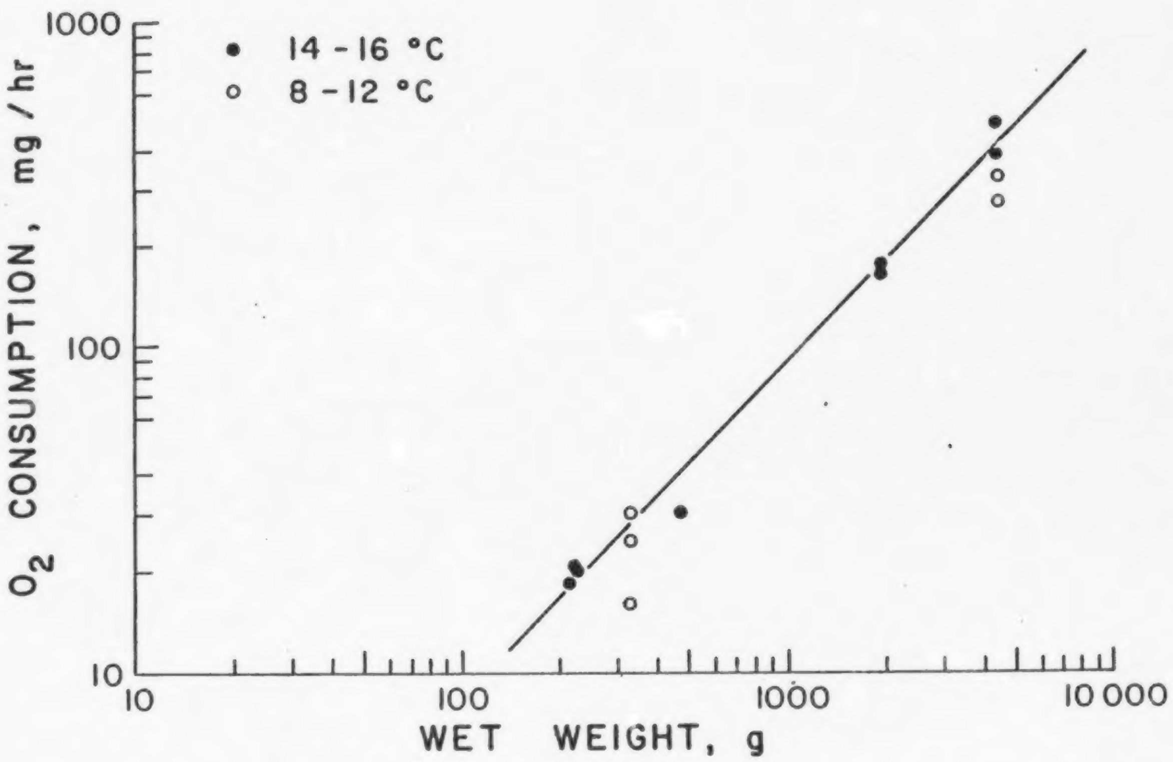


Figure 44. Relationship of oxygen consumption (mgO_2/hr/fish) to wet weight (g) for striped bass subadults and adults at two temperature ranges. The line is the best fit of data at 14-16^{o}C, where oxygen consumption = $0.072\ W^{1.029}$ (n = 8; r = 0.995).

TABLE 34. A. DAILY FOOD CONSUMPTION LEVELS FOR STRIPED BASS FED TO SATIATION AT DIFFERENT AMBIENT SEA TEMPERATURES

B. EVACUATION RATES FOR STRIPED BASS FED DAILY TO SATIATION

A.

Weight Range (g)	No. in Group	Temperature Range (°C)	Food Type	Daily Consumption Levels (% body weight) Range	Average	No. of Observations
Striped bass						
93-112	7	16-22	gelatin-squid	1.7-9.6	3.3	17
108-124	7	15-18	gelatin-squid	1.1-5.3	2.5	12
116-182	6	18-21	cut squid	6.1-17.1	9.4	10
152-286	20	16-22	squid, clam	3.8-12.8	5.3	25
410-797	14	16-22	squid, fish	1.4-7.9	4.6	17
482-868	14	16-20	squid, clam	1.1-8.5	4.5	23
563-1042	14	5-16	squid	0.8-7.2	2.9	21
1950	1	17-21	squid	0.4-9.6	3.0	23
1950	1	14-17	squid	1.6-5.3	3.6	11
4400	1	17-21	menhaden	0.3-3.7	1.7	29
4400	1	14-17	menhaden	0.5-5.2	2.2	11
1942-12500	38	18-22	squid, menhaden	0.4-3.9	1.7	8
1474-12500	39	8-15	squid, menhaden	2.4-5.2	3.7	10

B.

Weight Range (g)	No. in Group	Average Temperature During Test (°C)	Estimated 50% Evacuation Time (hrs) Using Bead Returns from each Test
1950	1	14	66
4400	1	14	54
4400	1	19	94,72,167
4400	1	21	49,46,42,34,65

(Table 34B), this feeding frequency could probably be extended to every other day. We calculated the caloric requirements at 14-16°C for an active one and four kilogram bass and determined that this energy demand can be met by consumption of about 3.5% menhaden or 8.5% squid of live body weight daily.

Variation in feeding habits has been reported for striped bass apparently depending on the availability of forage organisms. Hollis (1952) reported that during summer and fall (of 1936) the principal foods in saltwater areas of Chesapeake Bay were anchovy and menhaden and that during the winter months spot and croaker were dominant. In general, fish species dominated spring through summer to fall feeding, while invertebrate consumption increased during fall and winter (Stevens, 1966: Manooch, 1973). Bass fed mainly on menhaden in Narragansett Bay and on sand lance in Block Island Sound (Oviatt, 1977). Reduction in feeding by adults has been noted during spring and early summer. This is probably related to spawning activities (Hollis, 1952; Stevens, 1966).

The density recommended in Table 33 is based on successful rearing of adults in our flow through sea water system during the course of this study. The 2.4 g/l rate given is slightly less than the maximum density we actually maintained. The density recommended should offer insurance against stress especially at times of high oxygen demand. Oxygen requirements of adult bass are greater than those of sub-adults (Figure 44) on a per fish basis. On the basis of oxygen requirements, the density should probably remain 2 g/l or less.

Another factor influencing density in a culture situation is ammonia excretion. Typical rates for adult bass measured 48 hours after their last meal in sea water are shown below.

TABLE 35. EXCRETION RATES TYPICAL FOR ADULT STRIPED BASS AT TWO TEMPERATURES AT 30 o/oo.

Bass Weight (g)	Temperature (°C)	Ammonia Excretion ($N-NH_3$ mg/kg/day)
962	20.0	371.9
1950	14.5	247.2
4400	14.5	62.4 98.4

The amount of ammonia-nitrogen excreted per day by a one kilogram bass is about one-sixth the oxygen consumed by that bass during a day. Thus, oxygen levels are more critical for adults under culture conditions than possible ammonia accumulations.

Adult striped bass have few predators other than man. The diseases and parasites of adults are those presented in Table 24. These were discussed in detail at the end of Section 10.

Some of the environmental requirements associated with spawning can be inferred from the natural habitat section above and from the embryo section. These would include the temperature, salinity, pH, flow and other factors associated with successful spawning areas. In addition to freshwater hatchery spawning described by Bayless (1972) and Bonn et al. (1976), several attempts have been made to spawn adults in the laboratory.

Lasker (1974) described conditions under which a single successful spawning occurred in March in laboratory tanks. This occurred at 17°C in 50% sea water with a 15 hour light photoperiod cycle. Hormone dosages had been administered to this spawner.

We tried several approaches to spawning adults in the laboratory. Our lack of spawning success was not due to lack of development on the part of prospective broodfish, but rather to mechanical or weather difficulties. The conditions under which bass could be spawned in the laboratory require temperature, salinity, and photoperiod control together with sufficient food during the pre-maturation period of four to eight months before the desired spawning time. The following general procedures are recommended based on our studies.

Since the adults spend the majority of their prespawning feeding periods in coastal water, the best procedure would make use of natural photoperiod and ambient sea temperatures. The easiest case then would be to have the prospective broodfish spawn near their natural spawning times (e.g. in the mid-Atlantic region of April-May). To insure a greater measure of success the adults should be captured the preceding summer, allowing at least two-three weeks for adjustment to culture systems and initiation of active feeding.

The temperature of our ambient sea water generally falls in September from 20 to 16°C, in October from 15 to 10°C and in November from 10 to 5°C (Figure 3). These are ideal feeding temperatures corresponding to the natural fall feeding migrations along the Atlantic coast. During this period the broodfish designate should receive daily satiation feedings of diets both palatable and high in protein and lipid. Suggested diets are discussed below. For optimum feeding conditions, culture water should be 15-30 o/oo. The adult bass feeding frequency drops when temperatures fall below 4-5°C. It is important that adults consume enough to provide not only for daily maintenance, growth and activity, but also for gonad maturation during the period of temperature decline to 5°C. Three months is considered minimum especially for the larger females, which probably spawned the previous year expending their energy reserves that must be replenished. Although the adults are not actively feeding below about 4-5°C, food should be presented weekly and anytime the water temperature rises above 5°C. During the month of December and January, the sea temperature drops down to 2-3°C. This is cold enough and a period of 2-3 weeks at this temperature should provide the adults with their winter cue.

During this period the gonads are maturing, utilizing nutrients stored during the fall feeding period.

About the first of February the photoperiod should be extended a few minutes each day. More important is the gradual rise in temperature and decrease in salinity during the next few months to arrive at about 5-10 o/oo and 10-15°C in April. When the temperature is at about 12-15°C and the salinity about 7-10 o/oo, the broodfish should be anesthetized and checked for ripeness as described below under handling. After recovery the temperature-salinity levels should be maintained for a few days before reducing the salinity to 5 o/oo and raising the temperature to 16°C. If hormone injections are to be made, the best time would be at the time of checking the anesthetized broodfish. The procedures then follow those outlined by Bayless (1972) or Bishop (1975) and described below. Although it has been suggested that bass on the spawning grounds are fasting, it is wise under culture conditions to offer food to the adults when the temperature rises above 5°C even as the time of prospective spawning approaches. After spawning occurs, it is easiest to remove the fertilized eggs to their rearing system (see Section 8).

If two spawnings a year are desired, two groups of broodfish should be maintained under conditions similar to that described above. Each should be well fed after spawning for a number of months before beginning the cycle again. This insures full recovery of the fish. One group could easily be maintained in a system in a greenhouse to take advantage of natural photoperiod. The second group, if necessary, would have to be maintained under artificial photoperiod.

CULTURE METHODOLOGY

Capture Methods

In freshwater, mature bass have been successfully captured using electrofishing methods, bow and hoop nets, gill nets, hook and line, and traps (Bonn et al., 1976). We have obtained mature bass caught in gill nets (300' X 6' monofilament sink net with 6" bar mesh), by hook and line and through a local fish trap company from marine waters. Gill-netting is not a capture method usually chosen for taking fish in good condition. However, we used this method successfully during February and March. Success in obtaining living mature bass from stocks overwintering in Rhode Island was due to the low water temperatures and to the fact that the nets were fished for only a few hours at a time. Hook and line caught bass are usually taken in good condition, if plans are made beforehand so that a live well or other aerated tank is available to handle the fish. Most of our mature bass were obtained from a trap fishery. This was a successful method of obtaining large numbers of bass at once in good condition, especially if we went along to handle and choose fish. When choosing mature bass, size (see Table 28) and condition are the most useful tools. Ritchie (1965) proposed a biopsy technique to determine sex in the field from live bass. He evaluated the effects of this technique from returns of bass tagged and released in Chesapeake Bay (Ritchie, 1970).

Several methods are available to obtain mature bass in spawning condition. One is to catch both males and females during the normal spawning period in rivers and in sea water. This is the approach we followed in 1974 and 1975 in studying eggs and larvae on the Nanticoke River, Maryland. It is the procedure practiced at the established hatcheries at Weldon, North Carolina and Moncks Corner, South Carolina. Ripe broodfish captured in marine waters off Long Island during May-June, 1975 were successfully spawned.* A second approach is to catch probable broodfish during late winter and hold them for spawning. The Moncks Corner hatchery also utilizes this method. A third method, and one used at the Edenton National Fish Hatchery, North Carolina, is to catch and hold adults year-round in freshwater for spawning during the normal season. This method also applies to holding year-round in sea-water and varying salinity during natural spawning season as we have done. The fourth method is to catch and hold adults year-round for out-of-season spawning. This method has met with only partial success when tried by Lasker (1974) and by ourselves.

Post-Capture Handling

Adults should immediately be returned to aerated water of equivalent temperature and salinity to that of capture. The fish should be out of water as short a time as possible and handled gently to insure best chances of survival.

Transportation

Bonn et al. (1976) suggest transporting broodfish captured in freshwater in tanks equipped with mechanical aeration, supplemental oxygen and 0.3 to 1.0% salt (NaCl) solution. The density during transport they suggest is less than 45 pounds per 80 gallons (1 kg per 16 liters).

We successfully transported adults in tanks under quinaldine sedation in sea water with pure oxygen aeration. The use of pure oxygen is essential to reduce stress and ensure ease in oxygen consumption among the adults stressed by transporting and handling. Our density during transportation using the anesthetic could be as great as 1 kg per 4 l without losses.

Handling Procedures

Careful handling of adults as with the obviously more delicate larval stages is important. Adults can be moved, weighed and lengthed, observed and counted as easily as juveniles and sub-adults when proper methods are used. Whenever possible, it is wise to anesthetize the bass before handling. This need not be a full narcotic dose, but enough to slow the large bass down, in order to reduce stress during handling. We generally used quinaldine for anesthetizing the adults primarily because it was quick acting and used relatively small quantities in the large volume tanks holding these fish. During weighing of adults they were fully anesthetized with quinaldine at the rate of 0.02 ml per 1 in 500 1 adult anesthetizing tank containing rearing water. The bass were transferred to this from their larger rearing tank after the volume was reduced and enough quinaldine was added to slow the fish down so that they could be quietly herded toward the anesthetizing tank.

---

* Dr. Robert Valenti, MULTI-Aquaculture Systems, Inc., Ammagasett, New York.

Among the handling procedures specifically recommended are the following: 1) if at all possible, move each fish by dipping it with a quantity of water from one container to another. We use large heavy-duty polyethylene bags to dip and move large bass or heavy duty stretchers made of polyethylene. The surface of the bag is smooth and unabrasive and while it is possible for spines to penetrate the bag we find that if enough water is moved with the fish this virtually never occurs. 2) Move one fish at a time. This avoids fish to fish contact which in this spiny bass may be injurious to both. 3) Avoid letting the fish touch hard surfaces such as boat decks, fishpens, etc. If fish do end up on the deck retrieve them with a plastic bag or wet bare hands. Touching fish with cloth gloved hands always results in serious injury to the fish. Rubber gloves give no protection against spines and become so slippery that handling fish soon becomes a matter similar to trying to pick up a wet cake of soap in the bathtub. 4) If at all possible do not use a net of any sort to handle fish out of water. Nets may be used to crowd fish into bags in the water. 5) It is preferable to handle rod and reel caught fish by the hook and line on which they were caught than to use a landing net to boat them. 6) We have had best success handling large fish when we have aerated their holding water with pure oxygen as soon as possible after capture. During the process of handling each fish, a check should be made of the general condition of the adult. If there are any with external lesions suspected of being caused by disease or parasites, the fish should be separated and treated using a bath or dip of formalin, or malachite green. If the external lesion is a cut or abraded area resulting from netting during capture, an application of straight iodine or mercurochrome until the area is "red" works to kill any bacteria on the surface and reduces the likelihood of infection.

Bayless (1972) described the handling procedure adopted for induced spawning of broodfish. Intra-muscular injections of 125 to 150 I.U. of choronic gonadotropin per pound are given to females during the spawning season, when they are checked for development before being released into the holding tanks. Multiple injections did not show any improvement in ovulation results. The females are checked 24-28 hours later by removing eggs with a small catheter tube through the urogenetial pore and spawning time estimated. Generally, the female is checked about 30 minutes prior to the estimated spawning time and thereafter every 30-45 minutes until ovulation is observed. The female is lightly anesthetized and manually stripped. The eggs are fertilized with milt from males held separately. Bishop (1975) reported injecting males at the rate of 50 to 75 I.U. of CG per pound to provide maximum milt production in his Tennessee hatchery. Texas Instruments (1977a) administered 275 to 300 I.U. of CG per kilogram for females and 110 to 165 I.U. of CG per kilogram for males. The males at their Hudson River hatchery received a hormone injection only if milt production appeared limited or if second use of the male was anticipated.

Bishop (1975) described a slight variation to this procedure used at the Tennessee hatchery. Both females and males received hormone injections prior to stocking in circular holding tanks, but they were allowed to spawn naturally, not stripped. The fertilized eggs were retained in the tanks and the adults released. This somewhat reduced the handling of broodfish. Spawning through adjustments of photoperiod, salinity and temperatures was discussed above in the environmental requirements section.

Maintenance Procedures

Culture vessels--

Hatchery broodfish holding systems are described by Bonn *et al*. (1976). The culture system we utilized year round is shown in Figure 45. This system could be run totally on ambient sea water, on flow through, on total recirculation through a rapid sand filter, or partially on both. The water level in the tank, which held about 45,000 l, was controlled after filling the tank by the height of the "stand-pipe" in the discharge well. The loose threaded coupling allowed the entire tank to be emptied, since the drain was below ground level. This system was outside and thus subject to natural photoperiod. A similar system of two tanks holding about 8,000 l was set up in a greenhouse for spawning purposes. The photoperiod in the smaller system was adjustable. Salinity adjustments were made to both systems by the addition of dechlorinated tap water. This was very important during winter-spring spawning attempts. The outside system was covered and insulated with hay bales during the winter to reduce wind-chill cooling. A cover during the summer helped to keep algal populations low within the system.

Stocking Density--

The density indicated in Table 33 or 2 g/l maximum is equal to 110 kg per 45,000 l. This could include, for example, 20 one kilogram bass, 15 two kilogram bass, 10 four kilogram bass and 2 ten kilogram bass, or a total of 110 kg for these 47 adult bass. Of course, as the weight of each bass increases, the number must be decreased to maintain the density. We exceeded this rate during our spawning attempts utilizing the smaller system in the greenhouse. However, this system had supplemental oxygen aeration. It is preferable to have several large females and males composing the broodstock, all definitely mature enough to spawn, rather than many adults of questionable maturation state.

Maintaining Water Quality--

Water quality monitoring for the adult holding system should include temperature, dissolved oxygen, and salinity in open flow systems. If the system is recirculating, ammonia and nitrate should be included. If changes are made to freshwater during spawning from a preferred marine system during the rest of the year, the quality of the freshwater should be checked first for chlorine, copper and other metals, and pH. If the concentrations of these are near toxic levels, the freshwater should be treated before it is used.

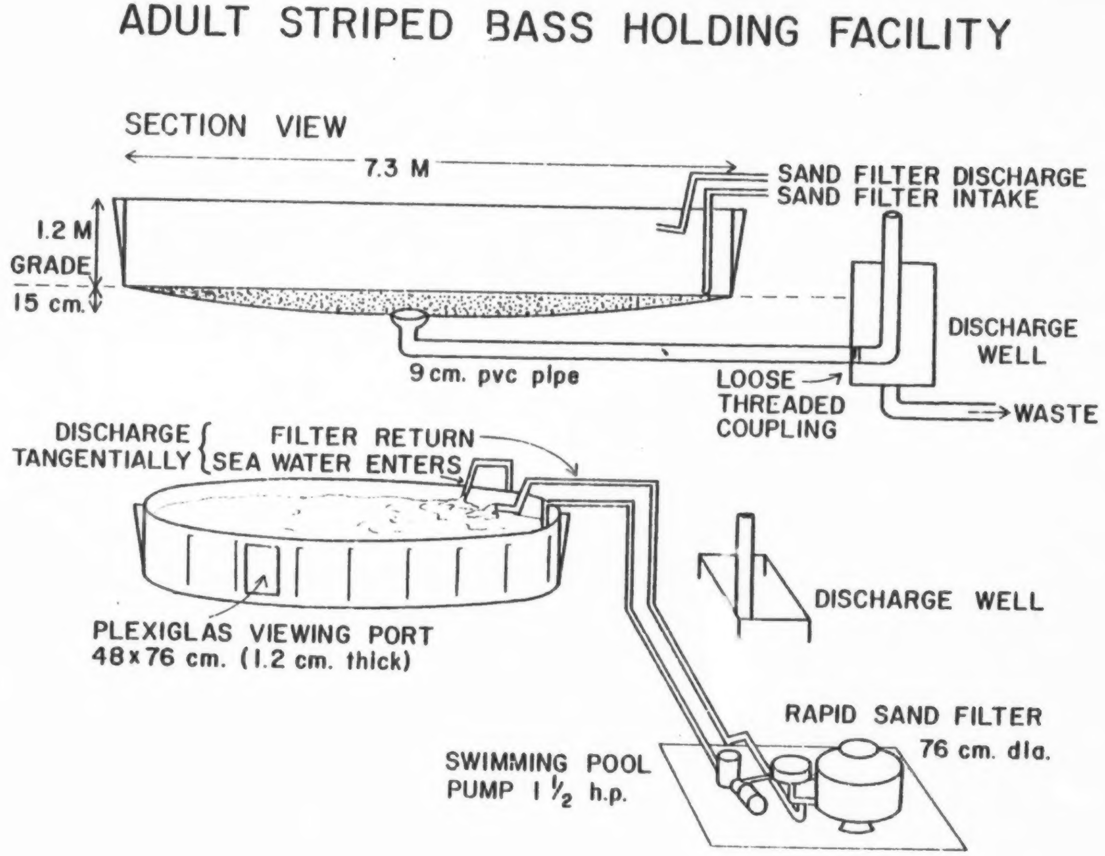

ADULT STRIPED BASS HOLDING FACILITY
SECTION VIEW
7.3 M
SAND FILTER DISCHARGE
SAND FILTER INTAKE
1.2 M
GRADE
15 cm.
DISCHARGE WELL
9 cm. pvc pipe
LOOSE THREADED COUPLING
WASTE
DISCHARGE TANGENTIALLY { FILTER RETURN SEA WATER ENTERS
DISCHARGE WELL
PLEXIGLAS VIEWING PORT 48x76 cm. (1.2 cm. thick)
RAPID SAND FILTER 76 cm. dia.
SWIMMING POOL PUMP 1 1/2 h.p.

Figure 45. Schematic of holding facility for adult striped bass.

Diet--

The food requirements for adults are outlined in Tables 33 and 34 and the abiotic factor section above. Among the acceptable diet items would be any of the naturally preyed upon species that were mentioned previously. Most of the fish species naturally consumed by adult bass average 15 to 19% protein on a wet weight basis and 1.0% (squid) to 16% (Clupea) lipid (Sidwell et al., 1974). Consumption of foods high in lipid appears to be essential for proper development and maturation of ovaries for successful spawning. It should be remembered that the eggs have a large oil glouble. For lipid to be stored therein, sufficient fatty acids must be present during the development process. Our spawning attempts strongly indicated that increased lipid content of the diet is important during the fall feeding prior to the onset of reduced feeding with winter temperatures. It seems that frequent satiation during this period is more important to gonad development for spawning than any feeding during the late winter-early spring just prior to spawning. The "spawn-not spawn decision" appears to be a function of the materials available for development and growth at least five to eight months prior to the spawning season.

Normal Conditions and Physiological State

Growth rates typically observed for adults at various temperatures in our ambient sea water systems are indicated below.

TABLE 36. GROWTH RATES (g/day wet weight) TYPICAL FOR ADULTS MAINTAINED IN SEAWATER AND FED DAILY

Weight Range (g) initial, final (mean)	No. in Group	Days in Period	Average Temperature (^{o}C)	Growth, (g/day) Per Group	Per Fish
576, 618	14+	20	19.6	2.10	0.15
618, 767	14+	34	18.4	4.38	0.31
767, 841	14+	55	11.2	1.35	0.10
2490, 3130	1	342	--	--	1.87
2948, 4520	1	342	--	--	4.59
6804, 8618	1	342	--	--	5.23
9752, 12470	1	342	--	--	7.95

+ See Figure 31 and Table 34.

These growth rates are approximately 0.5 to 1% of the bass weight when adjusted to the individual's weight. They are indicative of well fed bass under unstressful conditions.

We recorded activity of adults during daylight on movie film (8 mm). Swimming speeds of adults in our holding tank (Figure 45) ranged from 0.2 to 2 body lengths, or 12 to 60 cm/sec for the bass observed. Speeds were slower during quiet cruising periods and faster during active feeding periods.

General behavior of adults is similar to that described for subadults. Abnormal conditions, such as pugheadness, or disease and treatment, are as described for subadults (Tables 24 and 26).

SECTION 12

USE IN TOXICOLOGICAL RESEARCH

The striped bass is a desirable candidate for toxicological investigations in the United States for the following reasons. This species inhabits a wide range and is distributed along all three coasts (see Section 6). It is a commercially and recreationally important species (see Section 14) throughout its range. It is also an ecologically important member of the community it inhabits, not only along the coasts, but also within the coastal plain rivers. Its life stages are euryhaline and eurythermal, making them extremely useful in studies to determine sublethal differences in the physiology of toxicants over broad salinity and/or temperature ranges. In addition to being easy to culture, a great deal of the background research on this species has been reported.

To date, striped bass culture has been undertaken primarily by federal or state fish hatcheries to stock lakes, reservoirs and impoundments for sport fishing and shad (Dorosoma sp.) control. This work is done almost exclusively in fresh water, either in a hatchery or in ponds (Bonn et al., 1976). The culture methodology recommended in the previous sections for the life history stages of the striped bass, however, utilized sea water wherever possible. This was the case not only because of its availability and cost-effectiveness of use, but primarily to keep disease problems to a minimum. Although some fresh water (to reduce salinity) is needed during spawning and early larval stages, juveniles and adults feed and grow in sea water. For broodfish this feeding period is very important to successful production of gametes. We found that feeding is more voracious and disease problems reduced in sea water.

Using the methods recommended in earlier sections and summarized below striped bass life history stages can be cultured which are representative of the species. Figure 46 shows morphometric values of live juvenile bass seined from upper Chesapeake Bay rivers and of those reared in our laboratory. This is especially useful in terms of providing individuals for water quality and hazard evaluation testing, but has application to other areas of research.

CULTURE METHODS OUTLINED

The outline of recommended methods to secure batches of larvae or juveniles for toxicological studies that follows is based on the details presented in Sections 8-11. In general, if 20,000 prolarvae are required for studies, then a minimum of 40,000 eggs are needed. This estimate is based on 50% survival, although egg survival varies from 10-20% for artificially

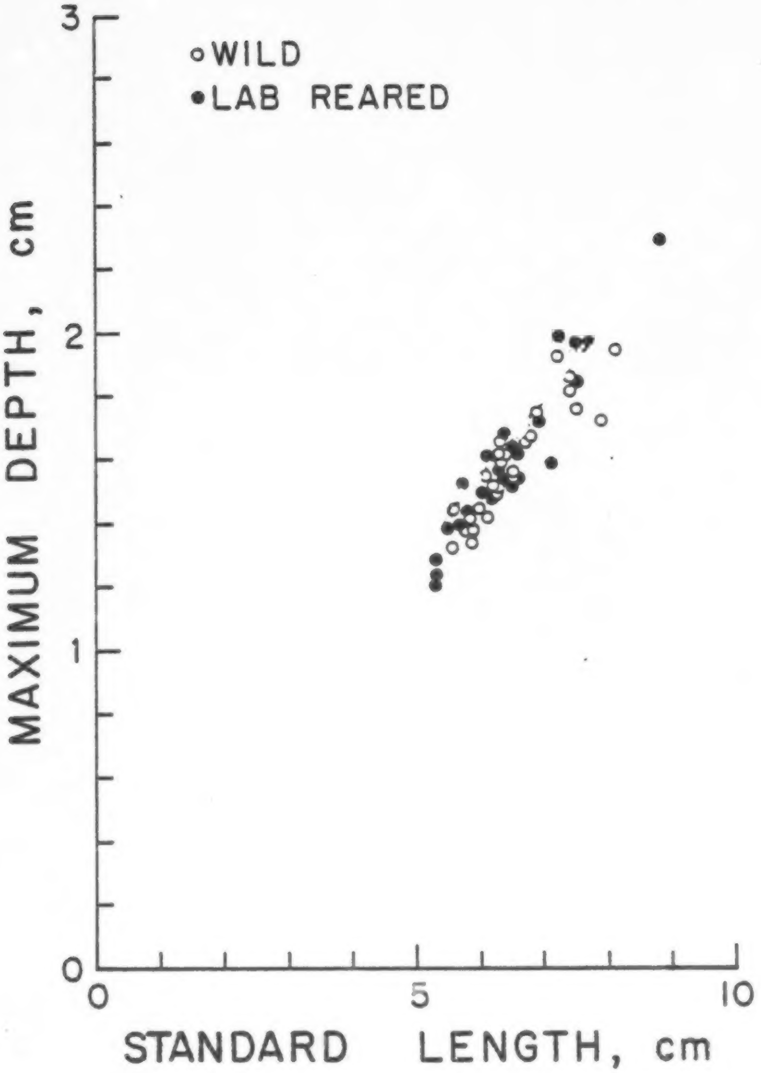


Figure 46. Measurements of maximum body depth (cm) and standard body length (cm) from live (anesthetized) striped bass seined from Maryland rivers (wild) or reared in the laboratory from eggs.

spawned to 60-80% for naturally spawned (see Table 32, Section 11) and fertilized eggs. This survival rate can be increased to 40-60% by using antibiotics in the rearing water (see Table 12, Section 8). If 2,000 postlarvae are required, then 2,500 prolarvae (80% survival through initial feeding; Figure 19, Section 9), or 5,000 eggs are required. These estimates are provided as a guide and may vary with broodstock, investigator, facilities and other variables.

A. Fertilized eggs can be obtained from natural or artificial spawnings.

  1) Artificially spawned eggs require maintenance of broodfish (mature adults) in culture system equiped with temperature, salinity, and photoperiod control with subsequent controlled spawning (see Section 11). Thus fertilized eggs may be obtained in the laboratory from spawning resident broodstock or from hatchery production by others.

  2) Naturally spawned eggs can be obtained easily by plankton or neuston net fishing in spawning rivers at the time of spawning (February-May).

  3) Collection of naturally spawned eggs insures genetic diversity not available among progeny of a mating under controlled spawning.

B. Handling of fertilized eggs to maximize survival and hatching.

  1) Eggs collected from plankton tows must be separated before transporting them to rearing containers.

    a) Raise salinity to 10 o/oo so eggs float and decant in water to shipping containers.

    b) Pack in shipping water up to 15-20,000 eggs per liter, or up to 200,000 eggs per styrofoam shipping box with bag liner about half filled with water and eggs.

    c) Shipping water should be at 5-10 o/oo or contain penicillin-streptomycin dose (50,000 IU-50 mg/1).

    d) Saturate shipping water and eggs with oxygen and fill space over water with oxygen.

    e) Secure bag tightly and cover box for transportation.

  2) Eggs secured from artificial spawnings can be stocked directly into rearing containers at a rate of approximately 100 per liter (i.e., up to 2,000 per McDonald hatching jar or 18-20,000 per 55 gallon drum recommended in Section 8). If eggs are obtained from a hatchery, they can be shipped as described above (b-e) at least 24 hours after fertilization and stocked into rearing containers as above.

  3) Handle eggs only in water, i.e., dip or pipette or siphon. Do not use dip nets.

4) When transferring shipped eggs to rearing containers check that water temperature of two are within 1°C of each other and that rearing water quality is optimum for egg survival (see Table 37).

5) Dead eggs, which float, should be removed daily from rearing containers while avoiding removal of hatched larvae.

6) Water quality, especially temperature, dissolved oxygen and salinity, should be monitored daily and maintained at optimum levels (see Table 37).

7) Fungus infection, which may occur in rearing water of < 5 o/oo, can be treated with a flush of malachite green (0.05 mg/l).

C. Handling larvae to maximize survival and growth.

1) Easiest method of securing prolarvae is from eggs on hand that hatch.

2) Prolarvae can be obtained from hatcheries

   a) Shipped as described above (B.1 b-e).

   b) Transferred from shipping box as described above (B3-5 except water quality should be optimum for prolarvae - see Table 37).

3) The recommended larval rearing system is static prepared tank system described in Section 9.(p. 94).

4) Larvae can be stocked at 100 per liter until actively feeding, when densities should be reduced to approximately 50 per liter.

5) Growing larvae should be graded to nearly equal size fish to reduce cannibalism.

6) The water quality in larval rearing containers should be monitored daily and maintained at optimum level (see Table 37).

7) Larvae should be fed at least twice daily beginning about 4-5 days after hatching an approved strain of brine shrimp nauplii and adults at rate of 50-100 nauplii per larvae at initiation of feeding and increasing to 300-400 nauplii-adults per larva by 15-20 days after hatching.

8) As larvae reach metamorphosis other foods such as ground squid or prepared diets (moist pellets) can be added to adult brine shrimp.

D. Handling juveniles.

1) Juveniles, if needed for research, can be reared from eggs and larvae, or collected by seining in spawning rivers.

TABLE 37. SUMMARY OF OPTIMAL REARING CONDITIONS FOR THE VARIOUS STRIPED BASS LIFE STAGES

	Eggs	Larvae		Juveniles & Subadults	Adults
		Prolarva	Postlarvae		
ABIOTIC FACTORS					
Temperature	16-20°C	16-21°C	18-22°C	>10 and <25°C	>10 and <24°C
Salinity	2-10 o/oo	5-15 o/oo	10-20 o/oo	10-30 o/oo	10-30 o/oo
Dissolved Oxygen	air saturated	air saturated		air saturated	air saturated
Light	natural photoperio	natural photoperiod		natural photoperiod	natural photoperiod
Turbidity	≤ 500 mg/l^a	≤ 100 mg/l^a		<4 mg/l^b	-
BIOTIC FACTORS					
Diet	not applicable	not applicable	15-20% body weight (dry) twice daily	5-8% body weight (wet) per day	3-5% body weight (wet) daily
Density	50-75 per liter	50-25 per liter	30-10 per liter	10-2 bass per 100 liters	2.4 g/l maximum (110 kg/45m^3)
Predators	many in natural habitat	many in natural habitat	canabalistic, many in natural habitat	some in natural habitat	none except man
Disease & Parasites	fungus	for summary see Table 24		for summary see Table 24	for summary see Table 24

a - fine grained sediments
b - bentonite

2) Juveniles collected from the field should be kept separate from any reared or other collected bass already in the culture system.

3) Juveniles can be seined as described in Section 10 (p. 124).

4) Water quality should be monitored daily and maintained at optimum conditions (Table 37).

5) Juveniles can be fed frozen brine shrimp, ground squid, prepared diets, or commercial trout feeds; the first is generally preferred.

Larval striped bass require large numbers of food organisms daily as shown earlier under diet requirements of this stage (Section 9). Although sufficient numbers are important to survival and growth through metamorphosis, the food organism must also be of a useful size. For a rearing system of moderate to large capacity, rearing large numbers of live organisms of varying sizes is suggested. The easiest method to provide this variety would be to grow brine shrimp (*Artemia* sp.). A number of very thorough procedures for hatching and growing are available. These include Nash (1973), Persoone and Sorgeloos (1975), Sorgeloos and Persoone (1975), Smith *et al*.(1978), Sorgeloos *et al*. (1978), and Beck *et al*. (1980).

Methods are also available for continuous culturing of algal species suitable as foods for the growing brine shrimp (Guillard, 1975; Persoone and Sorgeloos, 1976b). Yeasts or rice bran powder also provide a suitable diet for brine shrimp. We found culturing algae and brine shrimp in quantities sufficient to support larval populations of 10,000's to be relatively easy once the systems were set up. The problems we encountered in our early attempts were not of a technical nature, but rather simply not using large enough tanks for our continuous shrimp cultures. In a 12 ft. diameter pool, for example, an established population of brine shrimp under proper conditions (20-30°C, 30^{+} o/oo and algal or yeast foods) produces several kilograms of live food of various sizes daily.

We have recommended rearing larvae through metamorphosis at a minimum of 25-30% of their body weight (dry basis) daily on multi-sized live foods primarily to ensure successful survival and growth. Bengtson *et al*. (1978) have demonstrated that no artificial diet tested on the Atlantic silverside provided survival and growth equivalent to that provided by live *Artemia*. Care must be taken when formulating artificial diets for large scale or laboratory rearing to insure that levels of potentially harmful compounds are kept to a minimum. Commercially available trout and catfish diets have been reported to contain from 1 to 5 ppm chlorinated hydrocarbon residues (Branhn and Schoettger, 1975; Hansen *et al*., 1976). Other contaminants to avoid would be heavy metals. The importance of diet quality in toxicological research with fish was shown in studies reported by Mehrle *et al*. (1977).

In terms of research, a number of compounds have already been bioassayed using striped bass. In addition, reports of body burdens for heavy metals and xenobiotics, including pesticides and polychlorinated biphenyls, among natural stocks of striped bass are available as well as for our adults. These are discussed below.

PREVIOUS STUDIES

Bioassays

Embryos--

Toxicity data for striped bass eggs has been reported in the literature for copper, zinc and chlorine. O'Rear (1971) used 24-hour eggs per toxicant with the same water quality as in his larval experiments (Table 40). He found a 48-hour TLm of 1.85 (1.25-2.51) ppm zinc and 0.74 (0.605-1.73) ppm copper. Chlorine toxicity in flowing bioassay systems was reported for embryos by Middaugh et al. (1977), Morgan and Prince (1977) and Burton et al. (1979). The test conditions and water quality for these studies are included in Tables 40 and 41. Middaugh et al. (1977) exposed embryos continuously from 8 to 9 hours after fertilization till hatching. The percent hatch ranged from 56% for the control group (no chlorine exposure) to 0% for the embryos exposed to total residual chlorine concentrations of 0.21 mg/l. A general downward curvature of the vertebral column was observed among many of the larvae hatching after chlorine exposure. Morgan and Prince (1977) reported a 48 hour LC50 of 0.20 ppm chlorine for eggs exposed at less than 13 hours after fertilization and a 24-hour LC50 of 0.36 ppm chlorine for eggs exposed when more than 40 hours after fertilization. They observed blistering of the chorions among many eggs exposed to higher chlorine concentrations and noted generally slightly smaller larvae hatching from eggs exposed to chlorine concentrations. Burton et al. (1979) established the effects of interaction of total residual chlorine, change in temperature, and exposure time on survival of embryos.

Larvae and Juveniles--

The available information on the toxicity of certain substances to larval and juvenile bass is summarized from the literature in Tables 38 and 39, respectively. Table 40 summarizes the bioassay test conditions of each author, and Table 41 gives a summary of the water quality of their test water. Table 42 provides the analysis of the chemical substances used in these bioassays. While the majority of these bioassays have been on juveniles, the reports of actual tissue residues in bass (see below) have usually been reported for subadults and adults.

Larvae appear to be more sensitive than juveniles, based on available bioassay test results. Of the substances tested, larvae are least sensitive to chloride, potassium dichromate, and sulfate. Potassium dichromate has been recommended for the control of Monogenera and external protozoans in aquaria and ponds, respectively. Not included in Table 38 is a report by Hughes (1973) of 96 hour-100% survival in Instant Sea at 14,000 ppm (at Cl^-) and No Foam (Cresent Mfg. Co., Texas) at greater than 1,000 ppm.

Most of the substances tested have possible use in the pond culture (Bonn et al., 1976) of striped bass juveniles. Copper sulfate has been recommended in algal control. Casoron and Simazine have been recommended for control of aquatic vegetation, but the median tolerance limit of striped bass to Simazine is much lower than the rate recommended for control. Lindane and Malathion have been suggested, at a rate of 0.1-0.2 ppm and 0.5-1.0 ppm, respectively, in the control of parasitic copepods. Ethyl

TABLE 38. TOXICITY OF SUBSTANCES TO STRIPED BASS LARVAE[a]

Substance	96-hour TL_m (95% C.I.[b]) (mg/l)	Author
Acriflavine	5.0 (NA)	Hughes (1973)
Aldrin	0.01 (NA)	Hughes (1973)
Amifur	10.0 (NA)	Hughes (1973)
Butyl ester of 2,4-D	0.15 (NA)	Hughes (1971)
Cadmium	0.001 (NA)	Hughes (1973)
Chloride	1000 (NA)	Hughes (1973)
Chlorine	0.20 (NA)[e] 0.04-0.07 incipient	Morgan & Prince (1977) Middaugh et al. (1977)
Copper	0.05 (NA)	Hughes (1973)
Copper	0.31 (0.12-3.08)[c]	O'Rear (1971)
Copper sulfate	0.1 (NA)	Hughes (1971)
Dieldrin	0.001 (NA)	Hughes (1973)
Diquat	1.0 (NA)	Hughes (1973)
Diuron	0.5 (NA)	Hughes (1973)
Dylox	5.0 (NA)	Hughes (1971)
Ethyl parathion	2.0 (NA)	Hughes (1971)
Formaldehyde	10.0 (NA)	Hughes (1973)
HTH	0.5 (NA)	Hughes (1971)
Iron	4.0 (NA)	Hughes (1973)
Karmex	0.5 (NA)	Hughes (1971)
Malachite green	0.05 (NA)	Hughes (1973)
Methylene blue	1.0 (NA)	Hughes (1973)
Methyl parathion	5.0 (NA)	Hughes (1971)
Potassium dichromate	100 (NA)	Hughes (1971)
Potassium permanganate	1.0 (NA)	Hughes (1971)
Roccal	0.5 (NA)	Hughes (1973)
Rotenone	0.001 (NA)[d]	Hughes (1973)
Sulfate	250 (NA)	Hughes (1973)
Tad-Tox	5.0 (NA)	Hughes (1973)
Terramycin	50.0 (NA)[d]	Hughes (1973)
Zinc	0.1 (NA)	Hughes (1973)
Zinc	1.18 (0.25-2.46)[c]	O'Rear (1971)

[a] All 4-7 day-old larvae from Moncks Corner, South Carolina, tested at 21°C, except O'Rear (1971) which were tested in 14-19°C range, Morgan & Prince (1977) not specified, and Middaugh et al. (1977) at 18°C.

[b] NA = not available (i.e., neither given nor calculatable).

[c] 48-hour TL_m [d] 96-hour LC_0 [e] 24-hour TL_m

TABLE 39. TOXICITY OF SUBSTANCES TO JUVENILE STRIPED BASS

Substance	Test Temp (°C)	96-hour TL_m[a] (95% C.I.[b]) (mg/l)	Author
Abate	13	1.0 (NA)	Korn & Earnest (1974)
Achromycin	21-22	190 (153.2-235.6)	Kelley (1969)
Acriflavin	21	27.5 (NA) 16.0 (14.7-17.4)	Hughes (1973) Wellborn (1971)
Aldrin	13 21 20	0.0072 (0.0034-0.0152) LC_0 0.075 (NA) 0.010(NA)	Korn & Earnest (1974) Hughes (1973) Rehwoldt *et al*. (1977)
Amifur	21	LC_0 30.0 (NA)	Hughes (1973)
Ammonium hydroxide	15 23	1.9-2.85[c] 1.4-2.8[c]	Hazel *et al*. (1971)
Aquathol	21	610 (634-795)	Wellborn (1971)
Bayluscide	21	72 hr. 1.05 (0.94-1.18)	Wellborn (1971)
Benzene	17.4 16	10.9 ul/l (± 0.02) 5.8 ul/l	Meyerhoff (1975) Benville and Korn (1977)
Butyl ester of 2,4-D	21 20	3.0 (NA) 70.0 (NA)	Hughes (1971) Rehwoldt *et al*. (1977)
Cadmium	21	0.002 (NA)	Hughes (1973)
Carbaryl	17	1.0 (NA)	Korn & Earnest (1974)
Casoron	21	6,200 (5,210-7,378)	Wellborn (1971)
Chlordane	15	0.0118 (0.0057-0.024)	Korn & Earnest (1974)
Chloride	21	5000 (NA)	Hughes (1973)
Chlorine	18	0.04 incipient	Middaugh *et al*. (1977)
Cooling tower blowdown and power plant chemical discharge	4.5-6.0 18.5-26.0	>4.0X >4.0X [incipient LC_{50} without CL_2 3.6X (3.81X - 3.4X)]	Texas Instruments (1974b)
Co-Ral	21	62 (53-73)	Wellborn (1971)
Copper	21 17	0.05 (NA) 4.3 (NA)	Hughes (1973) Rehwoldt *et al*. (1971)
Copper sulfate	21 21-22 21	0.15 (NA) 0.6 (0.51-0.83) 0.62 (0.54-0.71)	Hughes (1971) Kelley (1969) Wellborn (1969)
Cutrine	21	0.1 (NA)	Hughes (1973)
DDD	17	0.0025 (0.0016-0.004)	Korn & Earnest (1974)
DDT	17	0.00053 (0.00038-0.00084)	Korn & Earnest (1974)
Dibrom	13	0.5 (0.1-2.4)	Korn & Earnest (1974)
Dieldrin	14 21	0.0197 (0.0098-0.00334) 0.25 (NA)	Korn & Earnest (1974) Hughes (1973)
Diquat	21 21	10.0 (NA) 80 (74-86)	Hughes (1973) Wellborn (1969)
Diuron (Karmex)	21	6.0 (NA)	Hughes (1973)
Dursban	13	0.00058 (0.00035-0.00097)	Korn & Earnest (1974)
Dylox	21	2.0 (NA) 5.2 (4.2-8.0)	Hughes (1971) Wellborn (1969)
Endosulfan	16	0.0001 (0.000048-0.00021)	Korn & Earnest (1974)
Endrin	17	0.000094 (0.000045-0.00019)	Korn & Earnest (1974)
E.P.N.	18	0.060 (0.025-0.150)	Korn & Earnest (1974)
Ethyl parathion	21 15	1.0 (NA) 0.0178 (0.0048-0.0657)	Hughes (1971) Korn & Earnest (1974)

a Unless specified otherwise

b NA = not available (i.e., neither given nor calculatable)

c Range of 96-hour TL_m in freshwater, 33% sea water, and sea water (95% C.I. given for percent mortality at 0, 40, 60, 80, and 100%).

(continued)

TABLE 39. (continued)

Substance	Test Temp (°C)	96-hour TL_m^a (95% C.I.^{b}) (mg/l)	Author
Fenthion	13	0.453 (0.216-0.955)	Korn & Earnest (1974)
Formaldehyde	21 21-22 21	15 (NA) 20 (15.4-26) 18 (10-32)	Hughes (1973) Kelley (1969) Wellborn (1969)
Heptachlor	13	0.003 (0.001-0.006)	Korn & Earnest (1974)
HTH	21	0.25 (NA)	Hughes (1971)
Instant Sea (as Cl)	21	LC_0 17000 (NA)	Hughes (1973)
Iron	21	6.0 (NA)	Hughes (1973)
Karmex (Diuron)	21	6.0 (NA) 3.1 (2.5-3.9)	Hughes (1971) Wellborn (1969)
Lindane	21 13	0.40 (0.35-0.46) 0.0073 (0.0045-0.0119)	Wellborn (1971) Korn & Earnest (1974)
Malachite green	21	0.2 (NA) 24 hr. 0.30 (0.27-0.33)	Hughes (1973) Wellborn (1971)
Malathion	21 13 20	0.24 (0.20-0.29) 0.014 (0.013-0.015) 0.039 (NA)	Wellborn (1971) Korn & Earnest (1974) Rehwoldt _et al_. (1977)
Methoxychlor	15	0.0033 (0.0021-0.0051)	Korn & Earnest (1974)
Methylene blue	21	12.0 (NA)	Hughes (1973)
Methyl parathion	21 13 20	4.5 (NA) 0.79 (0.17-1.40) 14.0 (NA)	Hughes (1971) Korn & Earnest (1974) Rehwoldt _et al_. (1977)
MS-222	21-22 22-28	31.5 (25.6-37.5) 24 hr. 50.0 (NA)	Kelley (1969) Tatum _et al_. (1965)
MS-222 with 20 o/oo	21-22	31.5 (26.6-37.5)	Kelley (1969)
Nickel	17	6.2 (NA)	Rehwoldt _et al_. (1971)
Oil field brine (as Cl)	21	LC_0 16600 (NA)	Hughes (1968)
Potassium dichromate	21	75 (NA)	Hughes (1971)
Potassium permanganate	21 21-22 21	4.0 (NA) 2.6 (2.17-3.12) 2.5 (2.1-2.9)	Hughes (1971) Kelley (1969) Wellborn (1969)
Polyotic	21	>1818 (NA)	Wellborn (1969)
PMA	21-22	1.1 (0.84-1.44)	Kelley (1969)
Quinaldine	21-22 22-28	4.5 (3.82-5.45) 24 hr. 22.0 (NA)	Kelley (1969) Tatum _et al_. (1965)
Quinaldine with 20 o/oo	21-22	5.0 (3.86-6.65)	Kelley (1969)
Reconstituted sea water	21-22	35 o/oo (NA)	Kelley (1969)
Roccal	21	1.5 (NA)	Hughes (1973)
Rotenone	21	LC_0 0.001 (NA)	Hughes (1973)
Simazine	21	0.25 (0.17-0.36)	Wellborn (1969)
Sodium nitrilotriacetic acid	20	5500 (NA)	Eisler _et al_. (1972)
Sulfate	21	3500 (NA)	Hughes (1973)
Syndet Ch	20	4.6 (NA)	Eisler _et al_. (1972)
Syndet Ga		8.7 (NA)	Eisler _et al_. (1972)
Tad-Tox	21	10.0 (NA)	Hughes (1973)
Terramycin	21 21-22 21	75.0 (NA) 170 (140.5-205.7) 178 (144-221) 165 (147-185)	Hughes (1973) Kelley (1969) Wellborn (1969) Wellborn (1971)
Toluene	16	7.3 ul/l	Benville & Korn (1977)
Toxaphene	17	0.0044 (0.002-0.009)	Korn & Earnest (1974)
m-xylene	16	9.2 (8.3-10) ul/l	Benville & Korn (1977)
Zinc	21 17	0.1 (NA) 6.7 (NA)	Hughes (1973) Rehwoldt _et al_. (1971)
2, 4, 5, T	20	14.6 (NA)	Rehwoldt _et al_. (1977)

TABLE 40. TESTING CONDITIONS FOR STRIPED BASS BIOASSAYS

Author	Size (mm) and Source of Fingerlings[+]	Volume in Test Container	No. Test Bass per Container	No. of Conc.	Testing Conditions
Burton *et al*. (1979)	eggs, larvae; B	206 ml	50	triplicates	flowing
Eisler *et al*. (1972)	mean 65; H	3 l	2.2 g/l	5 of 10 bass each + controls	static
Hazel *et al*. (1971)	20-93 TL; C	10 l	5-25 (generally 10)	2-5 + control	static
Hughes (1968, 1969, 1971, 1973)	30-50 TL; M	larvae: 1 l fingerlings: 2 l	10 2	10(?) + control	static
Kelley (1969)	60-80 TL; E	30 l	3 replications of 5 each	5	static
Korn & Earnest (1974)	14-83 SL; C	80 l	10 (<1 g/l)	-	flowing
Meyerhoff (1975)	mean 55; C	70 l	40	12 + controls	flowing
Middaugh *et al*. (1977)	eggs; mean 4.3, 6.7, 13.6 TL; M	eggs: 7 l larvae & juveniles: 4 l	eggs: 14, 9 ml aliquots of 603 ± 100 larvae: 20	eggs: 4 + controls larvae & juveniles: 7, 6, 7	flowing
Morgan & Prince (1977)	egg, larvae; M, V, P	1 l	-	4 + control with replicates (10)	flowing
Rehwoldt *et al*. (1971)	<200 TL; H	-	10	- + control	static
Tatum *et al*. (1965)	63-120; W	10 l	4	8 + control	static
Texas Instruments (1974)	40-100 SL; H (winter)	30 gal.	winter - 10 summer - 20	5 + control 6 + control	flowing
Wellborn (1969)	ave. 60 TL; E	40 l	10 (≤0.75g/l)	5-6 with 3 replications + 2 controls	static
Wellborn (1971)	ave. 47 TL; E	30 l	10 (≤0.4g/l)	several with 3 replications + control	static
O'Rear (1971)	4-7 day old larvae; M	3 l	40-137 (<0.3g/l)	6 + control	static

[+] C = California from Bureau of Reclamation, Tracy, California

E = Edenton National Fish Hatchery, North Carolina

H = Hudson River, New York

M = Moncks Corner, South Carolina Wildlife Resources Comm. Hatchery

W = Weldon Hatchery, North Carolina

V = Stannton River, Virginia

P = Potomac River, Maryland

B = Brookneal Striped Bass Hatchery, Brookneal, Virginia

TABLE 41. WATER QUALITY OF BIOASSAYS USING STRIPED BASS

Author	D.O. (mg/l)	pH	Salinity (o/oo)	Total Alkalinity (mg/l $CaCO_3$)	Total Hardness (mg/l $CaCO_3$)	Other
Burton *et al.* (1979) eggs	5.9	7.8	2.0	-	-	-
larvae	6.0	7.9	1.0	-	-	-
Eisler *et al.* (1972)	7.4	8.0	20	-	-	-
Hazel *et al.* (1971)	7.5	7.3-8.2	0-32	-	150-200	-
Hughes (1968, 1969, 1971, 1973)	-	-	Used reconstituted distilled water of 35 mg/l $CaSO_4$ & $MgSO_4$, 55 mg/l $NaHCO_3$, and 3 mg KCl			
Kelley (1969)	-	7.9-8.1	15,000-15,400 ohms/cm^2	59-62	32-36	Ca 3.9-4.1 ppm Fe_2 0.2 ppm Mg 2.0-2.2 ppm
Korn & Earnest (1974)	"satisfactory"	-	28-30	-	-	Turbidity 1-3 JTU
Meyerhoff (1975)	7.7	7.7	29	115	-	CO_2 6.0 ppm NO_3-N 1.5 ppm
Middaugh *et al.* (1977)	-	6.7-6.9	1-3	22	460	Ca 35.2 ppm Mg 90 ppm HCO_3 27 ppm SO_4 170 ppm
Morgan & Prince (1977)			Measured but not reported			
O'Rear (1971)	-	7.5-8.1	600-700 umhos/cm^2	140-200	110-150	Ca 60-80 ppm CO_2 6 ppm
Rehwoldt *et al.* (1971)	6.5	7.8	Hudson River water	-	53	-
(1977)	6.0	7.2		-	50	-
Tatum et al. (1965)	-	8.8	-	14	30	Cl 4.0 ppm NO_3-N 0.1 ppm
Texas Instruments (1974)	winter 11-8-14.0 summer 1.0-8.0	-	1.5-8.2 controls; up to 4.5	-	-	-
Wellborn (1969)	7.8	8.2	dechlorinated tap water	64	35	CO_2 3 ppm Fe_2 0.18 ppm
Wellborn (1971)	8.0	7.9	dechlorinated tap water	63	35	CO_2 2 ppm Fe_2 0.17 ppm

TABLE 42. ANALYSIS OF CHEMICAL SUBSTANCES USED IN STRIPED BASS BIOASSAYS

Substance	Grade or % of Active Ingredient	Active Ingredient
Abate	90%	0,0,0',0'-Tetramethyl 0, 0'-thiodi-p-phenylene phosphorothioate
Achromycin	250 mg capsules	Tetracycline hydrochloride
Acriflavine	Technical	Acriflavine (Hughes)
Acriflavine (neutral) NF	Technical	2,8-diamino-10-methylacridinium chloride and 2,8-diaminoacridine (Wellborn)
Aldrin	Technical	Hexachlorohexahydro-endo,exo-dimethanonaphthalene (Hughes)
	90%	1,2,3,4,10,10-Hexachloro-1,4,4a,5,8,8a-hexahydro-1,4-endo exo-4,8-ditnethanonaphthalene (Korn & Earnest)
Amifur	4.59%	Nitrofurazone
Ammonium hydroxide	-	Ammonium chloride
Aquathol	1.8 lbs/gal disodium salt of endothall	7-oxyalbicycle (2.2.1) heptane-2,3-dicarboxlic acid equivalent 15.5%
Bayluscide	5% heavy granular	2-aminoethanol salt of 2',5-dichloro-4'-nitrosalicylanilide
Benzene	Reagent	Benzene
2,4-D butyl ester	78%	Butyl ester of 2,4-dichlorophenoxy-acetic acid
Cadmium	Technical	Prepared from cadmium chloride
Carbaryl	98%	1-Naphtyl-N-methylcarbamate
Casoron	2% granules	2,6-dichlorobenzonitrile
Chlordane	60%	1,2,4,5,6,7,8,8-Octachloro-3a,4,7,7a-tetrahydro-4,7-methanoindan
Chloride	Technical	Prepared from sodium chloride
Chlorine	-	Chlorine gas (Burton et al.)
Chlorine	Analytical reagent	NaOCL (Middaugh et al.)
	-	Calcium hypochlorite (Morgan & Prince)
Cooling tower blowdown & power plant chemical discharge	1.0 X	Orthophosphate 1.5 ppm; Hydrazine 0.1 ppm; Cyclohexylamine 0.1 ppm; Lithium hydroxide 0.01 ppm; Boron 9.0 ppm; Potassium chromate 0.05 ppm as Cr^{+6}; Sodium hydroxide 0.03 ppm; Surfactant 1.0 ppm; Chlorine 0.1 ppm; Cl^- 5988 ppm; Na^+ 3400 ppm; SO_4^{-2} 890 ppm; Mg^{+2} 364 ppm; Ca^{+2} 164 ppm; K^{+4} 120 ppm; HCO_3^- 78 ppm; Si (as Na_2SiO_3) 8 ppm.
Co-Ral	25% wettable powder	0,0-diethyl 0-(3-chloro-4-methyl-2-oxo-(2H)-1benzopyran-7-yl)
Copper	Technical	Prepared from cupric chloride (Hughes)
	-	Copper nitrate (Reiwoldt et al.)
Copper sulfate	Technical	Copper sulfate
Cutrine	8.51%	Copper triethanolamine complex
DDD	99%	1,1-Dichloro-2,2,-bis(p-chlorophenyl1) ethane
DDT	77.2%	1,1,1-Trichloro-2,2-bis(p-chlorophenyl) ethane
Dibrom	90%	1,2-Dibromo-2,2-dichloroethyl dimethyl phosphate
Dieldrin	50%	Hexachloroepoxyoctahydro-endo, exo-dimethanonaphthalene (Hughes)
	85%	1,2,3,4,10,10-Hexachlor-6,7-epoxy 1,4,4a,5,6,7,8,8a-octahydro-endo-exo-1,4:5,8-dimethanonapthalene (Korn & Earnest)

(continued)

TABLE 42. (continued)

Substance	Grade or % of Active Ingredient	Active Ingredient
Diquat	35.3%	1,1-ethylene-2,2-dipyridilium dibromide (Hughes)
	3.73 lbs salt per gal; 2.0 lbs diquat cation	6,7-Dihydrodipyrido(1,2-a:2; 1'-c) pyrazidinium dibromide (Wellborn)
Diuron (Karmex)	80%	3-(3,4-dichlorophenyl)-1,1-dimethyl-urea
Dursban	99+%	0.0 Diethyl-0-3,5,6-Trichloro-2-pyridyl phosphorothioate
Dylox	80%	Dimethyl (2,2,2-trichlor-1-hydroxyethyl) phosphonate (Hughes)
	50% soluble powder	Dimethyl (2,2,2-trichloro-1-hydroxyethyl) phosphonate ester of butyric acid (Wellborn)
Endosulfan	-	6,7,8,9,10,10-Hexachloro-1,5,5a,6,9,9a-hexahydro-6,9-methano-2,4,3-benzodioxathiepin-3-oxide
Endrin	99%	1,2,3,4,10,10-Hexachloro-6,7-epoxy 1,4,4a,5,6,7,8,8a-octahydro-1,4-endo-endo-5,8-dimethanonaphthalene
E.P.N.	87.7%	0-Ethyl-0-p-nitrophenyl phenylphosphonothioic acid
Ethyl parathion	46.5%	0,0-diethyl P,p-nitrophenyl thiophosphate (Hughes)
	-	0,0-Diethyl-0-p-nitrophenyl phosphorothionate (Korn & Earnest)
Fenthion	-	0.0-Dimethyl-0-[4-(methylthio)-m-tolyl] phosphorothioate
Formaldehyde	37%	Formaldehyde (Hughes)
	Technical	37% formaldehyde gas solution (Kelley)
	Technical	Solution of 37%, by weight, of formaldehyde gas in water; 10-15% methanol added (Wellborn)

Substance	Grade of % of Active Ingredient	Active Ingredient
Heptachlor	99+%	1,4,5,6,7,8,8-Heptachloro-3a,4,7,7a-tetrahydro-4.7-methanoindene
HTH	70%	Calcium hypochlorite
Instant Sea	-	Prepared by Jungle Laboratories, Orlando, Florida
Iron	Technical	Prepared from ferrous chloride
Karmex (Diuron)	2.8 lbs diuron dibromide	3-(3,4-Dichlorophenyl1)-1,1-dimethyl-urea
Lindane	100%	1,2,3,4,5,6-Hexachloro-cyclohexane (Korn & Earnest)
	25% wettable powder	gamma isomer of BHC; 1,2,3,4,5,6-hexachlorocyclohexane (Wellborn)
Malachite green	Technical	Malachite green (Hughes)
Malachite green oxalate	Certified reagent, 96% total dye content	bis-(p-dimethylaminophenyl)phenyl-methane (Wellborn)
Malathion	95%	5,(1,2-dicarbethoxyethyl)0,0-dimethyl dithiophosphate ethyl phosphorodithioate (Korn & Earnest)
	25% wettable powder	0,0-dimethyl dithiophosphate of diethyl mercaptosuccinate (Wellborn)
Methoxychlor	89.5%	1,1,1-Trichloro-2,2-bis(p-methoxyphenyl) ethane
Methylene blue	Technical	Methylene blue
Methyl parathion	45%	0,0-dimethyl 0-p-nitrophenyl thiophosphate (Hughes)
	80%	0,0-dimethyl 0-p-nitrophenyl phosphorothioate (Korn & Earnest)

(continued)

TABLE 42. (continued)

Substance	Grade or % of Active Ingredient	Active Ingredient
MS-222	Practical	Ethyl-m-aminobenzoate
Nickel	-	Nickel nitrate
Oil field brine (as Cl)	-	-
Potassium dichromate	Technical	Potassium dichromate
Potassium permanganate	Technical	Potassium permanganate
Polyotic	10 gms active in 181.8 gms	Tetracycline hydrochloride
PMA	Technical	Pyridyl mercuric
Quinaldine	Technical	2-methylquinoline
Reconstituted sea water	-	Rila Marine Mix
Roccal	10%	Alkyl-dimethyl benzyammonium chloride
Rotenone	5%	Cube root
Simazine	80% wettable powder	2-chloro-4,6-bis(ethylamino)-*s*-triazine
Sodium nitrilotriacetic acid (NTA)	Monohydrated sodium salt	$(CH_2COONa)_3N \cdot H_2O$ (NTA)
Sulfate	Technical	Prepared from sodium sulfate
Tad-Tox	100%	Copper acetoarsenite (Prewitt-King Farms, Lonoke, Ark.)
Terramyacin	22.3%	Oxytetracycline HCL (Hughes)
	250 mg capsules	Oxtetracycline HCL (Kelley)
Terramycin, Globe Pet Tabs	50 mg active per tablet	Oxtetracycline hydrochloride (Wellborn, 1969)
Terramycin concentrate	25.6 gm/4 oz. soluble powder	Oxytetracycline hydrochloride (Wellborn, 1971)
Toxaphene	100%	Chlorinated camphene with 67-69% chlorine
Zinc	Technical	Prepared from zinc chloride (Hughes)
	-	Zinc nitrate (Rehwoldt *et al.*)

parathion has been used as a control for predators in ponds before the introduction of bass. HTH, a chlorine formulation, is used as a disinfectant in laboratories. Bayluscide can be used as a chemical control for snails that are known to act as the host for trematode parasites. Malachite green has been recommended for treatment of fungal, bacterial and parasitic infections in fishes. Its use is usually recommended at the rate of 1:15,000 for 15 to 30 seconds, or at 0.1 ppm as prolonged treatment. Polyotic has been recommended for bacterial control in fishes. It is usually used at the rate of 15 ppm as a prolonged bath, giving it a 10 fold safety margin for use on striped bass.

Not included in Table 39 is the report by Chadwick (1960) on the toxicity of Tricon Oil Spill Eradicator on juvenile (average 76 mm FL) striped bass from the San Joaquin River system. The fish tested showed no "distress" after 48 hours in 3.76 ml of Eradicator per 6,500 ml of river water (or a 0.005 percentage concentration). However, no survival was found after 10 hours in 7.5 ml of Eradicator per 7,500 ml of water (a 0.001 percentage concentration) and "distress" was observed at this concentration after an hour and a half of exposure at the test temperature of 65° F (18.3°C).

The ranges given in Table 39 for ammonium hydroxide median tolerance limits for juveniles are the spread reported by Hazel *et al.* (1971) for bioassays in fresh, brackish, and sea water at two temperatures. In fresh and brackish water (15°C) the toxicity of undissociated ammonium hydroxide was 2.8 ppm and in sea water the median tolerance limit was 2.0 ppm. At 23°C, the toxicity was 1.9 ppm in freshwater, 2.1 ppm in brackish water and 1.5 ppm in sea water. These authors observed that the striped bass tested were slightly more tolerant to undissociated ammonia in water of intermediate salinity than in either fresh or sea water. The bass also appeared less sensitive to the ammonia at 15°C than at 23°C.

Texas Instruments (1974) reported that behavioral changes related to acute toxicity of cooling tower blowdown and power plant chemical discharge to bass juveniles included hypersensitivity to the movement of an investigator in the laboratory, loss of equilibrium, and inability to regulate swimming attitude. Bass fed normally throughout chronic testing except those in 4.0x and 3.6x (1.0x contained 0.1 ppm chlorine) which fasted for 4 and 3 days, respectively, and then resumed normal feeding. New York University (1976) observed striped bass juveniles actively avoiding chlorinated discharge water (3.5°CΔT, 0.05 mg/l chlorine) when intake and discharge waters were mixed during behavior studies. Definite preference for quadrants of pure intake water was noted in counts at five-minute intervals in preference/avoidance chambers. Mortality and survival of larvae and juveniles after plume exposure at Indian Point during condenser chlorination was also determined.

Korn *et al.* (1976b) observed pronounced hyperactivity at high benzene levels (3.5 1/1). Bass exposed to the high level attempted to feed but were unable to locate and consume rations. Those exposed to the low benzene level had some success locating the food and about 50% was reported consumed. The control bass consumed all of their ration within five minutes.

By the end of the study (4 weeks) the control and low level groups were feeding normally, while the high level group consumed 50% of their ration.

The results of exposure to sublethal concentrations benzene (0, 5 and 10 ppm) at velocities of 7 and 14 cm/sec at 16°C in well water (0 o/oo) of striped bass 33-68 gm wet weight for 24-96 hours were reported by Brockson and Bailey (1974). The greatest increase in respiration rate of striped bass exposed to 5 ppm benzene was 45% after a 24-hour exposure, while the difference was least after a 48-hour exposure. The percent difference from controls in respiration rate at 5 ppm was consistently greater at 14 cm/sec velocities than at 7 cm/sec. The response to 10 ppm benzene exposure was very different. At both velocities the respiration rate decreased with exposure for 24 hours. The greatest depression in rate occurred at 7 cm/sec exposure for 48 hours. At 14 cm/sec, the respiration rate increased at exposure for 48, 72, and 96 hours. The standard metabolic rate determined for controls and then used to determine differences in respiratory rate of exposure to benzene was given as oxygen consumption (mg/kg/hr) = 214.154 - 1.798 wet weight (gm).

Courtois (1974) reported results of investigations on the sublethal effects of copper in freshwater and seawater on juvenile bass. Copper was found to modify osmotic balance; that is, saltwater acclimated bass exposed to copper dehydrated, while freshwater acclimated bass exposed to copper hydrated. Lower serum electrolyte concentrations (Na+ and K+) resulted in the hydrated state, and the dehydrated state produced elevated serum electrolyte levels. Courtois also determined that copper modified the osmotic balance of bass acclimated to different environmental temperatures and salinities. Actinomycin D (an inhibitor of Na+—K+ ATPase) and acetazolamide (a carbonic anhydrase inhibitor) were also shown to modify osmotic water balance in striped bass. A breakdown in osmoregulatory function was demonstrated at the gill membrane.

Dawson et al. (1977) reported the physiological response of juvenile striped bass exposed to 0.5, 2.5 and 5.0 ppb cadmium ($CdCl_2$) for 30-90 days and to 1.0, 5.0 and 10.0 ppb mercury ($HgCl_2$) in ambient seawater (22.6°C; 24 o/oo). Bass were allowed to recover in running seawater for 30 days after the longest exposure. Bass exposed to all cadmium concentrations for 30 days consumed significantly less oxygen (measured as gill-tissue consumption, $\mu l\ O_2$/hg/mg day weight) than did controls. Bass exposed for 90 days and those allowed to recover following the longest exposure respired at rates not significantly different from these of controls. The respiration rate of bass exposed to 1 ppb mercury did not differ significantly from that of controls, while those exposed to 5 ppm mercury for 30 days respired at a significantly lower rate than the control. After 60 days respiration of exposed and control mercury groups was about equal. The authors did not observe a significant change in the AAT or G6PdH activity in the livers of bass during exposure to cadmium or mercury. However, after a recovery period, those exposed to 5 ppb cadmium showed a highly significant decrease in both of these enzymes.

Tissue Residues

Concentrations--

Tissue residue concentrations of several metals found in waters supporting striped bass are summarized in Table 43. Arsenic residues from six adult striped bass taken from the Hudson River ranged from 0.23 to 0.67 ppm (Pakkala *et al*., 1972). The mean cadmium content of six Hudson River striped bass adults was 12.20 ppb (Lovett et al., 1972). Carpenter and Grant (1967) reported less than 10 μg/kg wet weight of cerium in edible portion. Windom *et al*. (1973) reported determining concentrations of copper, mercury and zinc from Savannah River striped bass of 2.5, 4.5, and 12 μg/gm dry weight, respectively. Alexander *et al*. (1973) analyzed forty-three bass taken off Montauk, New York, for mercury. They determined that striped bass over 5.7 kg wet weight would probably have mercury concentrations in muscle of greater than 0.5 mg/kg, while bass less than 3.2 kg would have less than 0.5 mg/kg mercury. Rehwoldt *et al*. (1978) reported average values of cadmium, lead, and mercury (mg/gm dry weight) for the Hudson River. Mercury content in musculature of seven bass from the Annapolis River, Canada in 1975 ranged from 0.11 to 0.43μg/g wet weight (Jessop and Doubleday, 1976). In 1976, 27 bass sampled from this river contained mercury ranging from 0.26 to 3.41 μg/g wet weight in flesh and from 0.01 to 1.78 μg/g wet weight in ovary samples (Jessop and Vithayasai, 1979).

A summary of chlorinated hydrocarbon concentrations in bass flesh and ova is presented in Tables 44 and 45 primarily from unpublished data supplied as indicated. Bischoff (1970) reported two striped bass from the American River, California, with PCB concentrations ranging from 2.15 to 2.52 ppm wet weight of flesh. The only report investigating the possible effect these chlorinated hydrocarbons might have on bass (specifically on reproductive success) is that of Boone (1973), which resulted in a number of as yet unanswered questions.

Depuration--

Korn *et al*. (1976a) investigated the uptake, distribution and depuration of C^{14} benzene in striped bass juveniles. Accumulation was greatest in the gallbladder, followed by mesenteric fat, colon, intestine, liver, brain, gill, heart, stomach and muscle tissue. Maximum concentrations were obtained in the tissues from 0.25 to 4 days after the start of exposure. Residues were depurated rapidly following termination of exposure. Gallbaldder, mesenteric fat, liver and gill maintained residues through the seventh day after exposure ended. Muscle tissue residues were undetectable 24 hours after exposure ceased.

Luhning (1973) anesthetized 12.7-20.3 cm striped bass in a 100 mg/l MS-222 solution (17.5°C) and found 57.9 μg/gm of MS-222 and 23.3 μg/gm *m*-aminobenzoic acid residues in muscle tissue immediately after a 30-minute exposure. Bass anesthetized with benzocaine (63.2 mg/l aqueous solution) contained 37.9 μg/gm free benzocaine and 1.4 μg/gm free p-aminobenzoic acid residues in muscle tissue following a 15-minute exposure. The esters and acids of both anesthetics decreased steadily with the length of recovery time. It appears that striped bass are the only species tested that can effectively hydrolyze the ester of MS-222 to m-aminobenzoic acid *in vivo*.

TABLE 43. RESIDUE CONCENTRATIONS OF HEAVY METALS REPORTED IN MUSCLE (FLESH) TISSUE FROM WILD STRIPED BASS

Area and Reference*	Metals (ppm wet weight)															
	Ag	As	Cd	Co	Cr	Cu	Hg	Mo	Mn	Ni	Pb	Sb	Se	Sn	V	Zn
Hudson River																
D (1)**	0.03	-	0.500	0.13	-	-	-	-	-	0.13	-	-	-	0.90	0.09	2.60
E (2)	-	-	0.249	0.409	0.613	2.80	0.105	-	0.265	1.45	-	-	-	-	-	5.49
Chesapeake Bay																
B (1)	0.003	0.250	0.03	-	5.000	0.350	0.350	-	-	1.000	0.500	<0.01	0.300	0.300	0.030	3.80
A (16)	0.026	2.020	0.055	-	0.237	0.320	0.052	0.000	0.167	0.256	0.268	0.000	0.916	0.526	0.155	3.46
A (10)	0.037	1.702	0.069	-	0.138	0.355	0.170	0.338	0.170	0.218	0.482	0.629	0.060	0.590	0.370	4.23
North Atlantic Coast																
A (10)	0.026	3.599	0.084	-	0.302	0.286	0.097	0.00	0.136	0.205	0.482	0.719	0.321	0.501	0.380	4.66
San Joaquin Delta																
A (5)	0.026	1.316	0.051	-	0.093	0.289	0.617	0.130	0.086	0.172	0.363	0.625	0.557	0.528	0.000	3.78
A (27)	0.028	1.981	0.068	-	0.051	0.305	0.432	0.187	0.112	0.188	0.387	0.587	0.494	0.461	0.200	3.73
A (7)	0.023	1.470	0.068	-	0.157	0.379	0.539	0.250	0.114	0.135	0.546	0.665	0.690	0.534	0.400	3.54
C (7)	-	-	0.150	-	0.330	2.300	0.330	-	-	-	1.300	-	-	-	-	31.5
Oregon Coast																
A (40)	0.028	2.289	0.062	-	0.151	0.204	0.858	0.130	0.111	0.181	0.442	0.604	0.464	0.460	0.315	4.23

* A = Hall et al. (1978)
B = Heit (1979
C = Kohlhorst (1973)
D = Tong et al. (1972)
E = Zawacki and Briggs (1976)

** Number in parenthesis is sample size for which mean values are given in table.

TABLE 44. SUMMARY OF HYDROCARBON RESIDUES REPORTED IN MUSCLE (FILET) OF STRIPED BASS

Area Sampled and Reference	Year Sampled	No. of Samples (fish)	DDE	DDD	DDT	Total DDT	Dieldrin	1016*	1254	1260	Total PCB
					mg/gm wet weight						
Shubenacadie River											
E	1976	18									tr-0.01+
Annapolis River											
E	1976	21									tr-0.09+
Rhode Island											
I	1979	8				0.01-1.3	<0.03*				0.04-14.1
Hudson River, NY											
B	1970	1	0.81	0.86	0.75	-	0.17				4.01
D	1973	22				0.72-9.83+					5.7-37.0+
H	1973		0.03	0.04	0.03		tr				
G	1975	2						0.56.4+			
G	1975	7							1.7-50.1+		
Atlantic Ocean off south shore L.I.											
G	1975	29						0	tr-3.59+		
Nanticoke River											
A	1972	2									2.7-4.1
Choptank River											
A	1972	2									4.7-5.7
Rappahannock River											
B	1970	1	0.16	0.19	0.13	-	0.02				0.56
San Joaquin Delta											
B	1970	1	0.45	0.21	0.23		0.02				0.99
F	1971	1	1.09	1.49	0.45		0.65				
C	1971	20									5.49-8.99+

* Includes 1016/1242 reported by Spagnoli and Skinner (1972)

+ Range of values given

A = Boone, Joseph. Fisheries Administration, Annapolis, MD.
B = Boyle (1970)
C = Curtis, T. California Dept. of Fish and Game, Stockton, CA
D = Harris, E. Rome Pollution Lab., N.Y. D.E.C. samples
E = Jessop and Vithayasai (1979)
F = Jones (1971)
G = Spagnoli and Skinner (1977)
H = Zawacki and Briggs (1976)
I = Authors data

TABLE 45. SUMMARY OF HYDROCARBON RESIDUES REPORTED IN STRIPED BASS OVARIES

Area Sampled and Reference	Year Sampled	No. of Samples	DDE	DDD	DDT	Total DDT	Dieldrin	Chlordane	1016	1254	1260	Total PCB
						mg/gm wet weight						
Shubenacadie River												
D	1976	7										0.01-0.10*
Annapolis River												
E	1975	18				0.005-6.67						0.08-12.8†
D	1976	26										0.01-8.50†
Rhode Island												
G	1979	6				0.09-14.98	<0.37	0.03-1.36				0.5-142.8
Hudson River												
B	1970	1	2.11	3.20	2.09	-	0.33					11.4
C	1973	1	1.90	0.62	0.67	-	0.42		NA	3.2	1.20	10.4
Nanticoke River												
A	1972	16	0.4-1.8	0.2-2.0	0.2-2.0	-	0.07-0.53 (n=11)	0.5-2.5 (n=7)				2.6-47.0
	1973	10	0.2-0.7	0.2-0.7	0.1-0.5	0.5-1.9		0.07-0.24				1.9-3.6
F	1974	5	0.4-1.9	0.3-1.3	0.2-1.0	-	0.03-0.13	-				2.3-10.5
Choptank River												
A	1972	4	0.20-1.40	0.37-1.60	0.36-2.00	0.93-4.70	0.16-0.34	(1.50) 1 fish				2.5-20.0
	1973	1	0.85	0.69	0.61	2.15	0.10			4.1		
F	1974	2	0.75,1.28	0.66,1.04	0.34,0.60	-	0.12,0.16					4.8,7.94
Rappahannock River												
B	1970	1	0.60	0.78	0.65		0.05					2.31
Roanoke River												
C	1973	5	0.39-0.93	0.13-0.24	0.13-0.73	-	0.01-0.07		NA	0.6-1.5	0.2-1.7	1.8-4.9
F	1974	2	0.75-0.89	0.46-0.70	0.54-0.59	-	0.04,0.21					2.9,3.6
Cooper River												
C	1973	5	1.14-5.01	0.45-2.12	0.83-4.00	-	0.02-0.11	0.10-0.45	NA	1.3-3.1	0.6-1.7	-
San Joaquin Delta												
B	1970	1	3.66	2.47	2.92	-	0.18					17.0

* Range unless only or two samples

A = Joseph Boone, Fisheries Administration, Annapolis, MD
B = Boyle (1970)
C = L. Glenn McBay, US Dept. of Interior, USFWS, Brunswick, GA
D = Jessop and Doubleday (1976)
E = Jessop and Vithayasai (1979)
F = Striper's Unlimited, No. Attleboro, MA
G = Authors data

Sills and Harman (1971) reported that residue levels in muscle tissue of striped bass exposed to 40 ppm of quinaldine at 4°C for 10 minutes reached 1.44-2.60 ppm, but were below 0.01 ppm 24 hours after end of exposure.

SECTION 13

POPULATION AND STOCKS

STRUCTURE

Sex Ratio

The females appear to spend more of their time offshore or at least in coastal waters. Holland and Yelverton (1973) found only 11.8% males in 1970-1971 in trawl samples off North Carolina. Vladykov and Wallace (1952) reported that 55% of the Chesapeake Bay population of striped bass sampled from commercial fishermen from June to January 1936-1937 were male. They observed a similar ratio for samples taken in Virginia and North Carolina, as did Scofield (1931) in California during 1927-1929. Age-sex ratio results of 852 striped bass sampled from commercial catches in Maryland waters of Chesapeake Bay in November-December 1976 showed that males accounted for 50, 44, 53 and 43% of the age I, II, III, and IV bass, respectively (Kohlenstein, 1980). Sex ratios by age class in the Hudson River during the 1976 spawning season showed dominance of age III and V males and age VII females (McFadden, 1977a). Schaefer (1968a) reported 14.3% males in populations sampled from 27 April to 24 November 1964, inhabiting the surf along the south shore of Long Island. Morgan and Gerlach (1950) observed that a greater percentage of the Coos Bay commercial catch was male during mid-April and May through late June of 1950. During late April to mid-May, females predominated in the catch. Merriman (1941) reported finding less than 10% males among bass sampled from Long Island and New England waters during 1936-1937. Sampling of sport and commercial catches in Rhode Island waters during 1973-1975 by present authors revealed 10.7% males. The largest male observed was 84.5 cm FL, while Schaefer (1968b) collected a nine year old 85 cm FL male during 1964.

Males apparently dominate on the spawning grounds when adult abundance is high. Kohlenstein (1980) analyzed the commercial landings during March-April on the Potomac River for 1966-1972 to estimate the sex composition of the spring catch. He calculated that the proportion (by numbers of fish) that were male was about 87% for ages III and IV, 73% for age V, 26% for age VI, and only 15% for age VII of the bass caught. Wilson et al. (1976) reported that the male to female ratio during the 1975 season was 3.44:1, while during 1974 the ratio was 4.15:1 on the Potomac River. Boynton* found a ratio of 0.2:1 during the 1977 Potomac spawning season.

Age Composition

Population studies in the St. John River, Canada, during 1971 and 1972 (Williamson, 1974) revealed greater than 20% age 4, 5, and 6 bass among the males (N=47) and age 5 and 7 among the females (N=149) sampled. Results from the same study in the Annapolis River, Canada, revealed greater than 30% age 4 and 5 males (N=54) and greater than 20% age 4 and 5 females (N=55).

Sampling in Maine waters during 1964 and 1965 by Davis (1966) showed predominance of the 1961 year class (4 year olds in 1965) and numerous bass of the 1958 year class. Schaefer (1968a), sampling the south shore of Long Island in 1962 and 1963, found a dominance of the 1958 year class (4 and 5 year olds, respectively) in the catches. Ages of bass ranged from 2 to 18 years in these samples. He noted that the 1958 year class was being replaced by the 1961 year class (2 year olds) in the October and November 1963 samples. Samples in 1964 of 168 large striped bass also showed the 1958 year class (6 year olds) dominant but with most of the bass between 4 and 7 years old. Commercial catches in northern waters (Long Island and New England) were dominated by 2 year olds in 1936 and by 2 and 3 year olds in 1937 (Merriman, 1941).

Tiller (1950) sampled commercial pound net catches from Maryland waters of Chesapeake Bay from October 1941 to November 1945. During this period the 1940 spawned bass dominated the catch in 1942 and 1943 (2 and 3 year olds) and continued to make up a significant portion of the catch in 1944 and 1945. The 1942 year class made a considerable contribution during the fall of 1943 and the entire year of 1944 (2 year olds). Most of the commercial catch in the Potomac River in 1962 were bass ages 2 and 3, while the upper Chesapeake Bay showed predominance of 2, 3, and 4 year olds (Nichols, 1962). Angler catches in the Potomac River during 1959-1961 were composed predominantly of age II (47.7%-85.3% of total) and age III (Frisbie and Ritchie, 1963). Age II fish made up 86.0% of the total catches for the 1960 sport fishing survey in the lower Patuxent estuary (Shearer _et al_., 1962).

Grant and Joseph (1969) determined the age composition in the James, York, and Rappahannock Rivers during June 1967-March 1968 from commercial and sport caught samples. The York and Rappahannock Rivers were dominated by 1966 year class, while the James River showed dominance of the 1965 year class during the sampling period. Grant (1974) sampled pound and fyke net catches for the age composition in these Virginia rivers from July 1967 through June 1971. He found seasonal changes in age composition slight with the older, migratory bass occurring more frequently in winter and spring catches. In each of the four sampling periods, age groups I-III (yearling through 3 years old) contributed over 84% of the catch from these rivers. During the 1969 and 1970 winter gill-net fishery sampling in the Rappahannock River, the 1966 year class was dominant (Grant _et al_., 1971).

---

* Walter Boynton (University of Maryland, Chesapeake Biological Laboratory, Solomons, Md.), "Spawning stock characteristics of striped bass in the Potomac Estuary," presented at the American Fisheries Society Annual Meeting held at the University of Rhode Island, Kingston, 22 August 1978.

The age composition of the commercial catch from Albemarle Sound, North Carolina, in 1962 was 95% ages 2 and 3, and only 4% in age 4 or older (Nichols, 1962). Trent and Hassler (1968) found the dominant age groups were III and IV for males and IV and V for females from Roanoke River gill net catches during the springs of 1963, 1964, and 1965.

Approximately 85 and 77% of the population of the Ogeechee and Savannah Rivers, Georgia, were reportedly composed of striped bass less than four years of age (Smith, 1970). Both rivers included strong classes of two and three year olds. However, young-of-the-year did not contribute significantly in the Savannah River but did in the Ogeechee River for the 1967 and 1970 sampling period. Scofield (1931) reported that most of the females caught in the commercial catch during 1927-1928 in California were 5 year olds with 6, 7, 4, and 8 year olds following in order of abundance. Additional age composition information can be found in Tables 28, 29, and 30 (p.148-150).

Size Composition and Growth Rates

Length frequency distributions have been provided by Radtke (1966), Schaefer (1968a), Tiller (1950), Vladykov and Wallace (1952), and Williamson (1974) in addition to those summarized in Table 30 . This table presents a comparison of growth in length for the ages specified and indicates the general growth rates for different areas of capture.

Rate of growth up to 70 cm can be computed from scales using the formula

$\ell = \frac{(L-1)\ell'}{L'} + 1$, where L = TL of bass, L' = radius of scale, ℓ = unknown TL,

and ℓ' = radius to annulus in question (Scofield, 1931; Merriman, 1941). Body length to scale radius relationships are available in Mansueti (1961), Robinson (1960), and Texas Instruments (1974a). Compensatory growth has been shown to occur in year 2 for striped bass from Chesapeake Bay and the Hudson River (Tiller, 1942) and in year 2 and 3 for bass from Albemarle Sound (Nicholson, 1964).

In Albemarle Sound, Trent (1962) found that the growth rate was almost linear among young-of-the-year striped bass (20-90 mm TL) from June to November. He calculated rates ranging from 0.272 to 0.433 mm/day for the five years of his study. In the Hudson River, Rathjen and Miller (1957) reported the greatest growth rate for young-of-the-year during June and July, continuing almost linearly to September-October. Texas Instruments (1975) observed essentially linear growth in length from July to November for young-of-the-year and an increase during April to July continuing almost linearly for yearlings. Vladykov and Wallace (1952) reported linear growth in length from April to August for bass beginning their second year. Scofield (1931) reported similar growth in length for young bass and 5, 6, and 7 year old females. The rate of growth in these locations was reduced during the winter months. Texas Instruments (1976) reported instantaneous growth rates (based on weight) calculated from young collected by beach seine during 1973, 1974, and 1975. The highest rates ranged from 0.0311 to 0.0407 for July-August, while the lowest ranged from 0.0145 to -0.0157 for October-November. Ware (1971) observed increasing growth in length among young-of-the-year from August to January and among yearlings from September through May. These bass had been stocked into freshwater lakes in Florida as four-day larvae.

Rathjen and Miller (1957) and Chadwick (1966) observed greater total length of young-of-the-year and yearlings in their samples taken in the lower Hudson River and in the lower Sacramento-San Joaquin Rivers, respectively. They proposed that these might have been slightly older bass that had moved downstream, or bass that had been feeding in the more productive areas of the rivers.

Growth is most rapid during these first years of life. This is the time when striped bass tend to remain in the rivers and estuaries near the site of spawning. Thus they are subject to changes in the environment of these moderately restricted water ways. A strong indication of density-dependent growth occurring in these nursery areas is seen in data presented by Austin and Hickey (1978).

Trent (1962) determined the linear relationship between standard, fork, and total length for bass 20-100 mm TL. These relationships are: $FL = 0.93835TL - 0.077817$; $SL = 0.80388TL + 0.55750$; and $SL = 0.85675FL + 1.22099$. Texas Instruments (1973) determined that $FL = 4.60 + 0.902TL$ for bass 103 to 667 mm, while Mansueti (1961) used the factor 0.93 to convert TL to FL, the factor 1.07 to convert FL to TL, and the factors 1.08 and 0.92 to convert SL to TL and TL to SL, respectively, for live bass. During the present study a linear regression for live bass of 12-65 mm as $FL = 1.55SL - 0.196$, and of 12-200 mm SL as $SL = 0.909FL - 1.805$ (see Section 10) was defined.

Length-weight relationships reported from different areas are given in Table 29 for different sexes, adult and young striped bass. Throughout their range it appears that after bass mature, the males of a given length weigh less than females of the same length (Merriman, 1941; Mansueti, 1961). Growth is more rapid during the second and third years of life, or before maturity, than in later years. Size at maturity for a number of stocks is presented in Table 28. Growth in length of females is greater after maturity than of males (Table 30). Graphic means of determining age and weight given the length of a bass are provided by Scofield (1932) and Clark (1938).

Condition factors (Kn) calculated by Trent (1962) ranged from 0.984 to 1.471 for bass 18.5-91 mm TL. Texas Instruments (1973) calculated condition factors (K) for young-of-the-year (25-100 mm TL) ranging from 0.94 to 1.25. Ware (1971) reported K-factors (Hile) varying between 1.31 and 2.79 for bass 76-483 mm TL from Florida lakes. He stated that the surviving bass with the K-factor of 1.31 was clearly emaciated, while those of at least 2.00 appeared very healthy. Values of K ranging from 1.658 to 2.540 for 0-450 mm SL bass were reported by Wigfall and Barkuloo (1976) for a Florida river system. Texas Instruments (1973) calculated condition factors (K) for 200-800 mm TL striped bass from the Hudson River and from Chesapeake Bay (using data in Mansueti, 1961). These factors ranged from 0.91 to 1.10 for the Hudson River bass and from 0.87 to 1.35 for Chesapeake Bay bass. Condition factors (K) for 37-70 cm FL adults on the spawning grounds of the Nanticoke River ranged from 1.06-1.63 (Westin, 1978). Three laboratory held adults (47-53 cm FL) were found to have condition factors of 1.16-1.26, while four migratory bass the same size showed factors ranging from 0.87 to 1.49.

ABUNDANCE AND DENSITY

Average Abundance

A model of the population dynamics of California striped bass is described by Sommani (1972). It is presented below with other models.

Population abundance based on Peterson mark-recapture estimates was 100,000 (1969-1973) for 5 year old males tagged in 1969 and about 150,000 during 1972-1973 for 5 year old males tagged in 1972 (Stevens, 1977a). The estimates were 5 million and 500,000 for tagged 3 year olds, respectively. Stevens (1977b) estimated a population index for 1958-1972 based on catch records. The estimated index was low in 1971 (86,020) and high in 1961 (322,250). He included a discussion of the biases of this index.

Texas Instruments (1974a) calculated two population estimates for the Hudson River during fall of 1973 using mark-recapture data. They estimated the population of young-of-the-year as 1,641,000 using Schumacher-Eschmeyer estimate and 1,680,000 using Peterson estimate. They discussed both briefly in relation to Hudson River striped bass. McFadden (1977a) reported Petersen estimates for 1974 and 1975 Hudson River young-of-the-year in late October as 1,288,000 and 1,024,000, respectively.

Austin and Hickey (1978) found an inverse relationship between the abundance of a year class in Chesapeake Bay and the modal length of age II+ bass in New York waters. In addition, they observed that the modal length of age III bass migrating into New York waters in the spring was a reliable index of the abundance of that year class. Observed modes rather than calculated modes for year classes 1954, and 1958-1962 resulted in more accurate estimates of New York landings for 1964 and 1965.

This study also provided reasonable data suggesting density-dependent growth of striped bass.

Changes in Abundance

Trent (1962) stated three factors - mortality, dispersion, and gear selectivity - presumed responsible (separately or in combination) for a reduction seen in young-of-the-year abundance as the season progressed. Sasaki (1966a+b) observed migrations of young-of-the-year and juvenile bass downstream from the Sacramento-San Joaquin Delta probably in response to food supply and/or water velocity changes. The survival and distribution of young bass were clearly defined by functions of water flow in this Delta system and abundance was greatest in the low salinity zone (Turner and Chadwick, 1972). Possible mechanisms for these relationships were discussed by the authors. Hallowing Point Field Station (1976) data indicated a probable relationship between short term river flow prior to spawning and juvenile abundance in the Potomac River. Conte *et al.* (1979) observed that the densities of bass eggs increased from ebb to flood tide and were greatest during flood tide at the mouth of the Sassafras River, Maryland. They observed from replicate tow samples taken during a tidal cycle on April 21st that larval densities showed a trend about opposite that of eggs. They felt that the short-term variations in these surface abundances were partly due to changes in the vertical mixing velocities and turbulence related to tidal currents. Using Hudson River data from Texas Instruments and New York University, McFadden (1977a) reported that a significant influence on the abundance of juveniles resulted from variables of predation, egg production, and rate of temperature change over the interval of 16-20°C from multiple regression analysis.

Average Density

Table 46 summarizes average densities of striped bass eggs, larvae, and juveniles as reported throughout their range. No attempt has been made to reduce these to a common basis. Eggs were found to vary in mean density from 3.5 to 17.0/m^3 during a series of replicate tows from 1900-2400 on April 21, 1976, at a single site in Upper Chesapeake Bay (Conte *et al.*, 1979). Mean densities of larval bass ranged from 0.9 to 15.9/m^3 at this site during the sampling period. Densities of yearlings and older striped bass in the Hudson River were reported from beach seine collections made in 1965-1974 (Texas Instruments, 1977b).

In general densities of eggs are highest from mid-water and bottom collections. Yolk sac and post-yolk sac larvae are densest near the bottom in day samples, but night sampling suggests vertical dispersion. Juveniles appear densest among bottom samples and from shore zone areas. This shorezone abundance declines in late fall and winter, but yearlings reappear in shore zone and bottom areas in spring. Tidal fluctuations appear to be unrelated to juvenile abundance in the shore zone from either day or night sampling.

TABLE 46. EXAMPLES OF ANNUAL DENSITIES OF STRIPED BASS REPORTED FOR DIFFERENT AREAS

Area	Year	Sampling Gear	Range in Mean Abundance or Density for eggs	larvae	juveniles	Reporting Units	Reference
Hudson River	1966-1967-1968	plankton net	0-10	0-12	-	#/1000 cu ft	Carlson & McCann (1969)
	1965-1968	semi balloon trawl	-	-	3.4-10.5 on bottom 1.3-22.3 off bottom	#/tow	
	1972-1975	beach seine	-	-	0-180	catch/unit effort	Texas Instruments (1976c)
	1969-1975	beach seine	-	-	0-60	catch/unit effort	McFadden (1977)
	1965-1975	beach seine	-	-	1.1-29.4	catch/unit area	McFadden (1977)
	1973-1974	beach seine, bottom trawl	-	-	0-232,0-12	catch/unit effort	Texas Instruments (1975a)
	1973	bottom trawl	-	-	0-75	#/10 min trawl	Lawler, et al. (1974)
	1973	epibenthic sled, tucker trawl	0-2500	0-546	0-45	$\#/1000\ m^3$	Texas Instruments (1976c)
		beach seine	-	-	0-240	catch/unit effort	Texas Instruments (1976c)
		bottom trawl	-	-	0-6	catch/unit effort	Texas Instruments (1976c)
	1974	epibenthic sled, tucker trawl	0-98	0-68	-	$\#/1000\ m^3$	Texas Instruments (1977b)
		beach seine	-	-	0-13	catch/unit effort	Texas Instruments (1977b)
Susquehanna River	1968-1969	seine, trawl	-	-	11-16	#/hectare	Carter (1973)
Potomac River	1973	plankton net	0-2100	0-6400	-	$\#/1000\ m^3$	Ecological Analysis Inc & Johns Hopkins Univ. (1974)
	1974	plankton net	0-13,833	0-600	-	$\#/1000\ m^3$	
	1975	plankton net	0-2480	0-869	0-51	$\#/1000\ m^3$	Hallowing Pt. Field Sta. (1976)
Chesapeake Bay	1954-1975	beach seine	-	-	0-29	#/haul	Boone*
James, York, and Rappahannock Rivers	1966-1967	30 ft balloon trawl	-	-	25-450	catch/trawl hour	Grant & Merriner (1971)
	1967-1972	minnow seine	-	-	1-6	catch/haul	Merriner & Hoagman (1973)
Albemarle Sound	1955-1961	18 ft balloon trawl	-	-	0-1.65	#/trawling min	Trent (1962)
Sacramento-San Joaquin Delta	1949		0-1193	0-773	0-88	catch/100,000 cu ft	Erkkila et al. (1950)
	1951	tow net on skis	-	-	0-3.5	#/1000 cu ft	Calhoun (1953b)
	1956-1959	beach seine	-	-	0-600	#/haul	Chadwick (1964)
	1957-1962	beach seine	-	-	0-350	catch/tow	Chadwick (1964)
	1963	otter trawl	-	-	0-700	#/10 min tow	Sasaki (1966)
	1959-1970	tow net on skis	-	-	34-116	index of abundance when pop. reaches mean length of 3.6 mm	Turner & Chadwick (1972)
	1970	tow net on skis	-	-	0-1293	#/acre ft.	Rogers & Stevens (1971)

*Joseph Boone, Fisheries Biologist, Maryland Dept. Fisheries Administration, Annapolis, Maryland.

Changes in Density

Downstream migration of juveniles during late summer and fall (Sasaki, 1966b; Texas Instruments, 1977b) reduces densities observed upstream. Changes in behavior of life history stages in response to tidal, diet, temperature or diel influences have been related to observed densities. For example, local movements of larvae, juveniles and yearlings have been well documented in areas of proposed power plants (Hudson River, Chesapeake-Delaware Canal and Potomac River) or pump storage and canal diversions (Sacramento-San Joaquin River valley) and examples from Hudson River studies (McFadden, 1977a) have been used to illustrate these changes. Yolk sac larvae are essentially planktonic but appear to concentrate near the bottom at night, dispersing somewhat during the day. Post-yolk sac larvae are capable of resisting currents and making directed movements. They exhibit nocturnal migration patterns strongly oriented toward the bottom. This orientation behavior appears to intensify as larvae approach the juvenile stage. Juvenile bass are first collected in mid-June to early July, depending on the time of spawning, from waters deeper than 6 meters. As water temperature increases, the juveniles migrate to shoal and shore zone areas. Falling water temperatures bring net downstream movement so that by December juveniles are generally absent from the shore zone, having either left the estuary or moved into deeper water for winter. Apparently, the abundance of juveniles in local areas is related to temperature, salinity, habitat type, diel patterns, and tidal stage. Comparisons of day/night beach seine catches in the Hudson River suggested movement into the shore zone at night, probably to feed or escape predation. Yearlings were found in deep water areas in early spring, throughout the estuary by summer. With falling water temperature, they moved into deeper water and downstream. Yearlings generally exhibit the same day/night pattern as juveniles, but appear less influenced by tidal fluctuations.

Pollution (e.g., siltation from dredging or runoff, heavy metals, chlorinated hydrocarbons, or temperature), dam building, and overfishing have been cited as factors contributing to striped bass stock depletions (Raney, 1952; Dovel and Edmunds, 1971). McHugh (1972) did not feel that the stocks were being depleted, at least not those represented by the New York landings from 1887-1970. He felt the increase in catch was caused by a real increase in abundance rather than an increase in fishing effort. This long-term trend in abundance was also apparent from the Virginia fishery landings (Grant, 1974).

NATALITY AND RECRUITMENT

Reproduction Rates

The number of eggs produced (i.e., fecundity) by the females of this species is highly correlated with weight, length and age. The number of eggs increases with age, although there is considerable variability between individuals of the same age group. An immature ovary contains small ova 0.07 to 0.125 mm in diameter. A mature ovary contains both small and large ova. The large ova average 0.22 to 0.76 mm in diameter, increasing to 1.0 to 1.35 mm at spawning. As they mature, the ova and ovaries change in color from cream to orange to pale, or grass, green. Fecundity data mainly from individual females, are plotted in Figure 43. Regression equations of fecundity (egg x 10^3) to body weight have been calculated for Hudson River (0.161 kg + 93.04; Texas Instruments, 1973), Roanoke River (75.9 kg [2.22] + 1.4; Lewis and Bonner, 1966), and offshore North Carolina (218 kg - 0.117; Holland and Yelverton, 1973) striped bass. These authors estimated fecundity at 173,000 eggs for Hudson River, 176,000 eggs for Roanoke River, and 318,000 eggs for offshore North Carolina striped bass, respectively, per kilogram of body weight.

Production rate estimates of eggs and larvae for three areas and several spawning seasons are summarized in Table 47. Survival rate from egg to yolk sac larvae determined from these estimates ranged from 1.6 to 190.0%. Survival rates for yolk sac to post-yolk sac larvae for 1974 and 1975 in the Potomac River were determined as 4.7 and 5.5%, respectively (Hallowing Point Field Station, 1976). Estimates of 1975 year-class survival rates in the Hudson River were calculated for four stages (McFadden, 1977a). The daily survival rates were 75.3% for egg to yolk sac larvae, 82.4% for yolk sac to post-yolk sac larvae, and 94.9% for post yolk sac larvae to juvenile.

Forecasting potential yields of striped bass from egg or larvae production estimates can be tenuous. The year class strength, or dominance, phenomenon of this species has received little attention until recently since Scofield's (1931) and Merriman's (1941) investigations. At the time of Raney's (1952) work there was "relatively little information available on probable conditions essential for the production of a good year-class" in striped bass stocks. Merriman and Scofield's observation that dominant year-classes are often produced by a comparatively small parental stock remains conjecture. Koo (1970), examining the commercial catch data from 1930-1966, concluded that the dominant year-class phenomenon was visible at 6-8 year intervals among Atlantic coast bass stocks. He felt it was a well-defined feature of the population dynamics of this species. Recently van Winkle *et al.* (1979b) reported periodicities of 20 years and 6-8 years which were neither simple nor predictable from times series analysis of the commercial catch data from 1930 to 1974.

Perhaps the major contribution since the early 1950's on the mechanisms of year-class strength for bass is the apparent agreement by investigators of the Hudson, Potomac and Sacramento-San Joaquin Rivers that control of population size is active within the first two months of life. This is a time of extreme vulnerability to environmental variation. A number of density-independent and density-dependent factors have been postulated with which to predict spawning success, or year-class strength. Low river flow and/or reduced run-off have been linked directly to reduced spawning activity and success (Hassler, 1958; Fish and McCoy, 1959), or to availability of

TABLE 47. ESTIMATES OF EGG AND LARVAL PRODUCTION AND SURVIVAL RATES

Area and Year	Egg Production	% Survival (calculated from productions)	Larvae Production (yolk sac)	Source of Data
Hudson River 1966	2.4×10^9	8.3	2.0×10^8	Carlson & McCann (1969)[a]
1967	0.52×10^9	36.5	1.9×10^8	Carlson & McCann (1969)[a]
1973	0.27×10^9	35.6	0.96×10^8	Texas Instruments (1975)[b]
1974	0.35×10^9	31.4	1.1×10^8	Texas Instruments (1975)[b]
Chesapeake & Delaware Canal				
1971	2.9×10^9	4.2	1.2×10^8	Johnson (1972)[a]
1973	9.5×10^9	7.3	6.9×10^8	Kernehan (1974)[a]
Potomac River 1973	5.0×10^8	190.0	9.5×10^8	Ecological Analysts & Johns Hopkins Univ. (1974)[a]
1974	3.8×10^9	2.9	1.1×10^8	Ecological Analysts & Johns Hopkins Univ. (1974)[a]
1974	4.5×10^9	1.6	0.7×10^8	Polgar *et al*. (1975)[c]
1974	4.5×10^9	1.6	0.7×10^8	Hallowing Point Field Station (1976)[c]
1975	9.9×10^9	5.0	4.9×10^8	Mihursky *et al*. (1974)[a]
1975	6.5×10^8	63.1	4.1×10^8	Hallowing Point Field Station (1976)[d]

[a] calculated and presented by Johns Hopkins University, 1975.

[b] Calculated as weekly standing crop for 5/15-eggs and 6/10-larvae.

[c] Estimates of fin fold larvae production given as 0.03×10^8.

[d] Estimates of fin fold larvae production given as 0.22×10^8.

larval food supplies (Polgar*). Low flow from water diversion has been directly related to year-class strength in the California stock (Turner and Chadwick, 1972; Chadwick _et al._, 1977; Stevens, 1977b). Winter water temperature in the spawning rivers has been linked directly to dominant year-classes (Merriman, 1941) or indirectly to year-class strength via effects on the larval food supply (Heinle _et al._, 1976). Additional effects of temperature and food availability on larval survival and growth have been demonstrated by Eldridge _et al_. (1977, 1980), Miller (1977), and Rogers and Westin (1979, 1980), while food supply in the nursery areas was considered an important factor by Austin and Hickey (1978).

Factors Affecting Reproduction

Although density-dependent factors have important affects on the reproduction and survival of striped bass, the density-independent factors are probably primarily responsible for variability in year-class strength (see Merriman, 1941; Koo, 1970; Heinle _et al._, 1976; Goodyear, 1978; van Winkle _et al._, 1979b). This also includes those factors over which man has some control, for example water diversions for irrigation or power plant cooling and water pollution by siltation or heavy metals. Most of the studies have examined factors affecting the survival of eggs and larvae with little information available specifically dealing with the physiological development of adults prior to spawning. Recently Wipple** suggested that egg viability could result from incompatibility of genetic combinations and/or affects of poor parental condition on gametes during maturation. Poor parental condition in this case was tied to chronic pollution levels, which were suggested to effect fecundity and egg viability depending on the degree of interaction of the parental genotype with the environmental stress. We have observed that mature adults require daily food consumption in excess of their growth and maintenance needs for at least three months prior to the decline in water temperatures to $5^{\circ}C$ (winter temperatures). It is during the two to three months at winter temperatures that the gametes develop from the excess energy stored during the active feeding period. If insufficient stores are available, gonad maturation ceases and reabsorption occurs (see Section 11). Results reported by Rogers (1978) and Rogers and Westin (1980) indicated that large females produced greater numbers of eggs with a greater dry weight. The benefit of this increased energy store is discussed together with the probable effects of temperature during the spawning season on the survival of the eggs and newly hatched larvae.

---

* Tibor Polgar (Martin Marietta Corp., Environmental Technology Center, Baltimore, Md.) "Factors influencing striped bass spawning success in the Potomac Estuary," presented at the American Fisheries Society Annual Meeting held at the University of Rhode Island, Kingston, 22 August 1978.

** Jeannette Wipple (NOAA Southwest Fisheries Center, Tiburon Laboratory, Tiburon, California). "The effect of inherent parental factors on gamete condition and viability in striped bass (_Morone saxatilis_)," presented at the Early Life History of Fish Symposium, Woods Hole, Mass., April 1979.

Recruitment

Based on age composition data the average recruitment into the fishable stock is at two or three years of age. A positive correlation between juvenile populations and subsequent commercial catch data, an indication of successful recruitment into the 3 to 6 years old class, has been shown for Maryland striped bass (John Hopkins University, 1976). Schaefer (1972) reported a statistically significant correlation between Maryland young-of-the-year data and New York landings. Austin and Hickey (1978) have demonstrated a more strongly correlated relationship between Chesapeake Bay young-of-the-year strength or age II + modal length of bass in New York waters and abundance (commercial harvest) in New York waters. Correlations between juvenile and adult stocks in the Sacramento-San Joaquin Delta have been demonstrated by Chadwick (1964) and Turner and Chadwick (1972). Stevens (1977a) observed that the Peterson method and indices from party boat catches appeared to be the best techniques of several investigated for monitoring three year old and older bass. He found that good correlation of Peterson estimates with young-of-the-year abundance indices or with river flows during the first summer of life were not available and discussed possible reasons for this. Stevens (1977b), however, calculated a recruitment index which he assumed measured 3 year old bass abundance based on sportfishing party-boat catch statistics. The analysis presented indicated that summer flows in the Sacramento-San Joaquin Rivers impact recruitment to the sport fishery several years later and are largely responsible for the fluctuations in population abundance.

The commercial yield-per-unit-effort for striped bass adults for 1965-1974 in the Hudson River was compared with the catch-per-unit-area index of juvenile abundance in July and August of the same year. A positive relationship occurred but adult abundance was not significantly related to early juvenile abundance (Texas Instruments, 1977b). Some evidence of the presence of compensation was suggested. McFadden (1977a) presented additional evidence in support of compensation for Hudson River striped bass. Chadwick et al. (1977) discussing factors regulating the striped bass population in the Sacramento-San Joaquin Delta mentioned compensatory processes, which until recently were considered dominant. However, now they feel that density-independent processes, especially the mortality of juvenile due to water diversions within the delta, play a major role in controlling the population size.

Determining recruitment as indicated by larval abundance has been hampered greatly by collecting techniques (John Hopkins University, 1976). Neither the number of eggs spawned nor the size of the adult population appears related to recruitment, although the factors affecting reproduction rates and success effect recruitment.

Although Chadwick (1969) stated that the relationship between parent stock and recruitment could not be defined from available population measurements for the California striped bass stock, Sommani (1972) demonstrated that a modified Ricker curve represented recruitment in this stock.

McFadden and Lawler (1977) have indicated that the Ricker stock-recruitment curve is applicable to the Hudson River striped bass population. However, analysis of their approach (Resource Management Associates, 1979) has shown that the Ricker model is not an accurate representation of the available stock-recruitment data for Hudson River striped bass. Goodyear (1978) concluded that this relationship is not known for any Atlantic Coast bass stocks. He suggested several models (i.e., Lawler, 1972; Van Winkle *et al.*, 1974; McFadden, 1977a; Christensen *et al.*, 1977) that have been proposed to portray this stock-recruitment relationship. These models are discussed in more detail below with the other models that have been developed to predict the population or stock behavior of this species.

MORTALITY

Mortality Rates

Rates of mortality among striped bass eggs and larvae are indicated in Table 47 from estimated productions using standing crop sampling data. Polgar (1977) has presented methods for estimating the mortality rate for successive bass life stages from egg to metamorphosis using field survey data. The methods assume either an uniform age distribution or an exponential age distribution within each stage. Using 1974 Potomac River field data, the calculated mortality rates for each stage for both methods are 2.35 for eggs, 0.32 for yolk-sac larvae, and 0.07-0.19 for the stages from yolk-sac absorption to metamorphosis.

Mortality rates in the striped bass population of the Sacramento-San Joaquin Rivers were calculated from disk dangler tag-returns for 1958-1962 (Chadwick, 1968), 1965-1971 (Miller, 1974), and 1958-1968 (Sommani, 1972). Table 48 summarizes the instantaneous mortality, exploitation and survival rates from these and other studies as available. The choice of tag was based primarily on an evaluation of five types reported by Chadwick (1963). The mortality rates calculated for Virginia rivers were based on returns from tagging studies using internal anchor tags. The rates calculated for North Carolina were from tagging studies using Floy dart tags. The calculated annual fishing mortality rate for bass tagged in the ocean off North Carolina and recaptured from North Carolina to Maine was 35% (Holland and Yelverton, 1973).

Chadwick (1968) stated that the tag returns gave a reasonable estimate of mortality rates, but that the exploitation rates were probably underestimated. Miller (1974) discussed the biases from the differences in returns by sex and size of estimates of this population parameter described from these studies. Decline in harvest rates (Table 48: 0.37 in 1958 to 0.12 or 0.096 in 1968) was attributed indirectly to decline in angler success which probably caused reduction in fishing effort (Miller, 1974). This may be related to a declining population.

Other studies evaluating tag types suitable for population dynamics evaluation were done by Bonner (1965), Davis (1959), and Lewis (1961). All three selected the streamer type.

TABLE 48. ESTIMATES OF SURVIVAL AND MORTALITY RATES FOR SOME STRIPED BASS STOCKS

Area	Year	Survival Rate		Exploitation Rate		Expectation of Death from Natural Causes*		Instantaneous Mortality Rate						Reference
								Total		Fishing		Natural		
California	1958	0.319[a]	0.316[b]	0.372[a]	0.372[b]	0.309[a]	0.312[b]	1.14[a]	1.15[b]	0.62[a]	0.63[b]	0.52[a]	0.53[b]	[a]Chadwick (1968)
	1959	0.534	0.535	0.247	0.255	0.219	0.210	0.63	0.63	0.33	0.34	0.30	0.28	
	1960	0.601	0.590	0.243	0.253	0.156	0.157	0.51	0.53	0.31	0.33	0.20	0.20	[b]Sommani (1972)
	1961	0.662	0.672	0.190	0.202	0.148	0.126	0.41	0.40	0.23	0.25	0.18	0.15	
	1962	0.592	0.675	0.200	0.200	0.208	0.126	0.52	0.39	0.25	0.24	0.27	0.15	[c]Miller (1974)
	1963	0.511	0.632	0.281	0.246	0.208	0.122	0.67	0.46	0.39	0.31	0.28	0.15	
	1964	0.557	0.705	0.235	0.167	0.208	0.128	0.67	0.35	0.36	0.20	0.31	0.15	
	1965	0.655[c]	0.664	0.142[c]	0.136	0.203[c]	0.200	0.42[c]	0.41	0.17[c]	0.17	0.25[c]	0.24	
	1966	0.628	0.678	0.179	0.176	0.193	0.147	0.46	0.39	0.22	0.21	0.24	0.18	
	1967	0.647	0.703	0.160	0.148	0.193	0.149	0.44	0.35	0.20	0.18	0.24	0.18	
	1968	0.687	0.750	0.120	0.096	0.193	0.154	0.37	0.29	0.14	0.11	0.23	0.18	
	1969	0.614		0.193		0.193		0.49		0.24		0.25		
	1970	0.688		0.119		0.193		0.37		0.14		0.23		
	1971	0.660		0.147		0.193		0.41		0.18		0.23		
North Carolina	1972							0.278		0.036		0.243		Holland and Yelverton (1973)
Virginia	1963	0.33[d]	—[e]					0.66[d]	—[e]					Grant & Merriner (1971)
[d]York River	1966	0.04	0.02					0.96	0.98					"
[e]Rappahannock River	1967	0.02	0.01					0.98	0.99					"
	1970	0.07	0.14					0.93	0.86					Merriner & Hoagman (1973)
	1971	0.02	0.15					0.98	0.85					"

*Chadwick (1968) ratio of 1962 to 1961 returns as valid estimate of 1961 survival, and annual expectation of death from natural causes from 1962-1964 was equal to 1958-1961 mean. Miller (1974) assumed expectation of natural death was equal to mean for 1959, 1960 and 1965.

Merriman (1941) estimated that natural mortality accounted for about one-third of the two year olds in 1936 which were not taken by the fishery. Chapotan and Sykes (1961) found fishing mortality particularly high during the first three years of life. Kohlenstein (1980) assumed that the natural mortality of three year old males ranged from 10 to 15% in the Potomac River stock. From tag returns he estimated the commercial and sportfishing harvest (i.e., fishing mortality) to be 27-42% in Maryland waters, where the landings showed over two-thirds of the commercial catch occurred during January-May. Using these estimates of natural and fishing mortality, the average age distribution of the commercial and sport catches, estimates that sport fishing lands about one-half as many as commercial fishing, he calculated the best estimates for age III male and female fishing mortality to be 35% and 4%, respectively.

Effects of changes in natural mortality rates upon striped bass populations using various models were discussed specifically by Chadwick (1969), Sommani (1972) and Saila and Lorda (1977). These and other models which include parameters to deal with density-dependent and density-independent mortality variations are described in more detail below. Arguments presented by Chadwick *et al*. (1977) suggested density-independent factors as the major influence on population size, while McFadden (1977b) suggested density dependent factors stabilize the population.

Factors Causing or Affecting Mortality

Predators are responsible for mortalities of egg, larval, and juvenile striped bass. The effect of reduced food availability on larval survival has been specifically discussed by Eldridge *et al*. (1977, 1980), Miller (1977), Rogers (1978), and Rogers and Westin (1979, 1980). Other factors such as cold winter water temperatures and ice scouring of marshes (Heinle *et al*., 1976) have been linked to larval survival through resulting food availability. Only rarely have parasites been directly related to mass mortalities of striped bass (Sniesko *et al*., 1964).

Striped bass are among the more dominant species in the area during their spawning. The larvae may, therefore, compete for food with white perch or shad larvae or croaker juveniles. Hollis (1967) included a section describing the food habits of brackish water fishes which have one or more of their important food items in common with young-of-the-year and yearling striped bass. Included in this group were white perch, johnny darter, bay anchovy, alewife, blueback herring, hickory shad, Atlantic menhaden, tidewater silverside, brindle shiner, silver minnow and golden shiner.

Many of the physical factors affecting mortality have been discussed recently by Chadwick *et al*. (1977). These include changes in river flow, whether by diversion or damming, entrainment and impingement in power plant cooling systems, and pollution. Pollution can include siltation from dredging or runoff, heavy metals, and chlorinated hydrocarbons. Implication of pollution in a specific case of large-scale mortality in California was discussed by Kohlhorst (1973).

DYNAMICS OF STOCK OR POPULATION

A number of mathematical models have been produced in an effort to predict striped bass population behavior in relation to environmental changes, especially those influenced by power plant cooling systems. The earliest models were life cycle models (Lawler, 1972; Sommani, 1972; van Winkle et al. 1974) or stock-recruitment models to aid in stock management (Chadwick, 1969; Sommani, 1972). Later models added the dimension of the spawning river hydrography to the biological observations of this species as well as elements to handle a wider range of possible factors affecting the stock. These models include U.S. Nuclear Regulatory Commission (1975), Lawler, Matuksy and Skelly Engineers (LMS, 1975), Polgar et al. (1975), Warsh (1975), Eraslan et al. (1976), and Christensen et al. (1977). There are several analyses and comparisons of these models or their elements (Wallace, 1975; van Winkle et al., 1976; Saila and Lorda, 1977; Swartzman et al., 1977; Resource Management Associates, 1979). Two comprehensive studies of the effects of power plant operation on the Hudson River with emphasis on striped bass population estimates, distribution and abundance have been completed by Barnthouse et al. (1977), and by McFadden (1977a) and McFadden and Lawler (1977). A brief description of these models and their analyses follows to give the reader at least a historical perspective.

Many of the models cited deal with the Hudson River striped bass stock and have been developed primarily to assist in estimating the impact of power plants along the river. Collection of hydrodynamic and biological data for this river and its aquatic populations was most intense during 1965-1975 for hearings on the Consolidated Edison of New York, Inc., proposed power plants at Indian Point in Buchanan, N.Y. The first model proposed (Lawler, 1972) simulated Hudson River striped bass life stages from egg through adults using a deterministic approach based on a set of differential equations similar to those employed to model transport processes in engineering and physics. Although it included parameters to predict cropping due to entrainment and impingement as well as natural mortality in young-of-the-year (YOY) stages, it included no parameters to simulate spatial relationships of YOY bass and river hydrodynamics. Models simulating tidal transport, YOY migration, and vertical hydrodynamic effects were included during 1972-1975. The resulting model, known as the Real-Time Life Cycle Model (RTLC), is described in detail in LMS (1975) and McFadden and Lawler (1977). It is essentially three submodels including a model of the hydrodynamic behavior of the Hudson River and models of the YOY and adult populations within the river. The dynamic nature of the hydrodynamic simulation provides the "real-time" aspect of the model. To simulate the interaction of the tidal action in the Hudson and the vertical diurnal migration of the larvae, the model divides the river into two layers of equal depth. The temporal variations in flow due to tidal action and migration of larvae are then simulated by evaluation of the hydrodynamic function and distribution of the organisms at three hour intervals. The YOY and adult models are linked by the YOY model which supply recruits to the adult model. Assuming natural survival rates that are density-independent, the adult generates an adult population (using Leslie matrix approach) with a known female ratio which supplies eggs to the YOY model.

In the Final Environmental Statement related to Indian Point No. 3, the Nuclear Regulatory Commission (USNRC, 1975) staff generated two striped bass simulation models (a YOY and life cycle model) to assist them in determining both the short-term impact on YOY bass of entrainment and impingement by power plants on the Hudson River and the long-term impact on the bass fishery and population structure. These two computer simulation models, and their programs, have also been presented separately in full detail by Eraslan et al. (1976) and van Winkle et al. (1974). The daily total-averaged, longitudinally cross-section-averaged (one dimension) YOY model includes contributions from tidal dispersion, convection, and migratory transport. The model's mathematical formulation is based on the concept of balancing instantaneous rates of change from convection, dispersion, migration, mortality, survival and growth, and entrainment and impingement of each of the six life stage populations (egg through juvenile III) within each discrete element of the river. The life-cycle population model is based on a Leslie deterministic, discrete-time scheme incorporating age-dependent fecundity and survival. It assumes a constant sex ratio and so the model deals only with adult females. Natural mortality is assumed to be independent of population size and to occur at all ages over age one. Fishing mortality is applied to the older bass and is varied with weight. The model allows estimates of the proportion of surviving YOY with and without density-dependent mechanisms as well as estimates for entrainment and impingement. Other factors affecting mortality incorporated into the model are cannibalism, and growth and survival related to food availability.

Another early life cycle model is that of Sommani (1972) for the Sacramento-San Joaquin striped bass stock. This model was formulated to ensure sound fishery management of the stock. Estimates of natural mortality rates were made from tag-recapture data, while the relationship of population size and recruitment was generated from catch per angler day data and observations of summer YOY abundance. To model the observed stock-recruitment relationship for the 1960-1965 data, Sommani investigated the functions of both Beverton-Holt and Ricker. He found that, although Chadwick (1969) suggested the reproduction curve for California bass followed the Ricker function, neither Beverton-Holt nor Ricker functions include environmental factors in spawner-recruit function improved the fit to the data. He concluded that a Ricker function modified by expressing the June-July outflow in the rivers was the best model of the population. This modification includes the effect of outflow on the production of young bass (substantiated after Sommani's study) involving spawning capacity and food availability outlined earlier in this section. Recently, Christensen et al. (1977) have developed a multi-age-class model based on broad biological principles. It is capable of producing stock-recruitment relationships similar to the classical Beverton-Holt and Ricker type curves. The model can be used to examine the potential impact of power plant cropping of YOY for the range of stock-progeny relationships generated.

Striped bass egg and larval simulation models for the Potomac River and the Chesapeake-Delaware Canal have been proposed by Warsh (1975). Each model consists of a series of differential equations describing the mass balance of water (vertically and longitudinally) and bass life stages within each segment of the estuary. This model includes terms describing changes in spawn density caused by advection, dispersion, spawning, natural mortality, entrainment, and larval behavior. This latter element is seen by Warsh as the most uncertain for the systems modeled. Polgar et al. (1975) reported analysis of the transport processes in the Potomac River to determine their role in the spatial and temporal history seen in ichthyoplankton distributions during 1974 spawning seasons. They used this analysis to develop empirical models to describe the distribution of eggs and larvae in this river. Recently, Wallace (1978) has shown that the data available from Hudson River field surveys indicates that larval stages of striped bass do not simply drift downstream like solute particles. She observed that these early life stages appeared able to maintain their longitudinal position in both fresh and brackish water areas of the estuary. Results generated from models that simply add larvae into a circulation pattern (i.e., USNRC, 1975) would, thus, she postulates, appear questionable in estimating the density of larvae at a power plant intake site.

Most of the models described above were developed as tools to assist decision-makers in assessing the impact of proposed power plants along rivers supporting striped bass spawning populations. During the course of this assessment dialogue, many of the assumptions and functions utilized in the models' development have been reviewed, compared and analyzed. A great deal of this review and analysis has been directed at Hudson River models, especially the first model and its offspring.

In one of the first comparisons of early Hudson River bass models, Wallace (1975) compared key assumptions of the simple and complex models developed to the field data available. She concluded that the Lawler (1972) model agreed with the data on more points than any of the other models investigated, which included the USNRC (1975) model as well as six other early models. She suggested that the state of verification of the models at that time favored the "one based on the fewest assumptions and on the broadest body of available data" to have the greatest predictive power. Her comparison also mentioned that the complex models disagreed on the inclusion (Lawler's) or exclusion (USNRC's) of compensation as a factor influencing the population.

The compensation function incorporated into the LMS (1975) Real Time Life Cycle (RTLC) model (i.e., the later form of the early Lawler complex model) was analysed by van Winkle et al. (1976) using two types of sensitivity analysis. They criticized the conceptual basis, especially "the lack of a sound biological basis for the left limb" of the function. They presented an alternative form for the compensation function which has a plateau extending from zero to some critical density. This is the form of compensation used in both the van Winkle et al. (1974) and Eralson et. al. (1976) models. They showed that this formulation had a sounder biological basis than the LMS compensation function. In a recent review and evaluation of the LMS/RTLC model, Resource Management Associates (RMA, 1979) included analysis of the two natural mortality models used in the LMS/RTLC. Both represent the compensation process. One natural mortality formulation is based on the Ricker model and the other is based on the Beverton-Holt model. This analysis points out that, although the most recent presentation of the LMS/RTLC model (McFadden and Lawler, 1977) shows the later to be the most statistically acceptable representation of the mortality rate, the former formulation was adopted. It also shows that the Ricker model is not an accurate representation of the available Hudson River stock-recruitment data. In addition to the review of this portion of the LMS/RTLC model, the RMA (1979) analysis stated that the model documentation presented no verification results or analyses for the model. It was found that the model did not accurately represent the observed variability in the YOY bass in the Hudson.

Another element utilized in several of the models is the Leslie matrix model. Saila and Lorda (1977) demonstrated its value as a tool used to examine the dynamics of the Hudson bass population. They examined the available data on survival rates of various YOY stages and performed sensitivity analysis on the effects of changes in these rates on short-term dynamics of that population. They simulated an increase in mortality of from 2 to 20% on five YOY stages from an equilibrium population, alone and in combination. They observed that simulated mortality increases to only one stage resulted in little change in the adult population, while increases in mortality at each stage resulted in reduction of the population. Also any reduction in the fishing mortality in one to several of the age-classes (3 to 20) of the population would permit a higher tolerance of additional YOY mortality in the model.

Swartzman _et_ _al_. (1977) compared eight simulation models used to assess the impact of power plants on important fish species. Seven of them were striped bass models of Hudson and Chesapeake stocks. All of these assumed that without power plant operation, the bass populations remained in equilibrium. They discussed the two hypotheses for the manner in which compensatory mortality acts and concluded that the striped bass data is not sufficient to infer the spawner-recruit relationship. Thus it is difficult to tell which hypothesis (LMS, 1975 vs. USNRC, 1975, or Van Winkle _et_ _al_., 1974 and Eraslan _et_ _al_., 1976) is more reasonable for striped bass. Swartzman _et_ _al_. (1977) found that the major differences in biological assumptions in the bass YOY models was the choice of stage durations and inclusion of compensatory mortality at both high and low fish densities. In the bass life cycle models, they observed that the major difference in the predictions of yield and population reduction resulted from using density-dependent or density-independent fishing mortality, and using different values for the probability of natural survival of bass in age classes 1 to 3. They concluded their review with suggestions for improved documentation of models and greater cooperation and exchange of modeling ideas.

THE POPULATION IN THE COMMUNITY AND THE ECOSYSTEM

Much of this information has been presented in other sections. The general features of the environment supporting the various life stages of striped bass can be found in Sections 8 to 11 in the subsections on natural habitat. Species composition and seasonal abundance in some communities which include bass have been reported for the surf zone by Schaefer (1967), for the Hudson River estuary by Perlmutter _et_ _al_. (1967a and b), Institute of Environmental Medicine (1977), Texas Instruments (1976c, 1977b), and McFadden (1977a), for the Delaware River estuary by Schuster (1959), and de Sylva et al. (1962), for the Chesapeake-Delaware Canal by Bason et al. (1975), for the Potomac River by Ecological Analysts and Johns Hopkins University (1974), Johns Hopkins University (1974), and Hallowing Point Field Station (1976), and for the Sacramento-San Joaquin Delta in Turner and Kelley (1966). Changes in environmental factors and the effect on the community and stock are discussed in these reports and in subsections above.

SECTION 14

EXPLOITATION AND MANAGEMENT

EXPLOITATION

Fishing Equipment

Gears --

Striped bass are landed using both commercial and sport fishing gear. The commercial gear presently employed are listed in Table 49. Changes in gear during the development of the fishery were discussed by Koo (1970), Raney (1952), Scofield (1931), and Vladykov and Wallace (1952). Rosko (1966) and Moss (1974) described sport fishing gear and techniques in detail.

Recent changes in contribution by each type of commercial gear is indicated in Table 50 by percentage of the total landed within each state. There has been an increase for all states in handlines, otter trawl and drift gill net catches and a decline in pound net and haul seine catches during the second five year period.

Boats --

A variety of vessels from small outboards to larger trawlers, depending on type of fishing gear, are used. Sport fishing utilizes party boats of various descriptions.

Fishing area

Geographic ranges --

Striped bass support sport or commercial fisheries throughout their distributional range (see Section 6). The relative abundance can be inferred from landings by state (Table 51) where the sale of bass is permitted.

Fishing for striped bass occurs in reservoirs, rivers, bays, coastal ponds and estuaries throughout its range. Fishing also occurs along exposed coast, sandy beaches, open headlands, offshore wrecks and banks generally within 5 to 10 miles (8 to 16 km) of the coast depending on the gear used and the legality within the state (see Table 51).

TABLE 49. SUMMARY OF INFORMATION AVAILABLE ON STRIPED BASS FISHERIES

State	Type of Exploitation	Equipment	Area	Season[a]
Maine	sport	handlines	Gulf of Maine & estuaries	spring & summer
New Hampshire	sport	handlines	Gulf of Maine	spring, summer & fall
Massachusetts	sport	handlines	coastal areas	spring & fall
Rhode Island	sport & commercial	handlines floating traps fixed gill net haul seine	coastal areas, ponds, & Narragansett Bay	spring & fall
Connecticut	sport	handlines		
New York	sport & commercial	handlines haul seine pound net otter trawl gill nets	coastal areas of Long Island, Hudson River[b]	spring & fall winter & spring
New Jersey	sport & commercial[c]	handlines fyke net haul seine pound net otter trawl	coastal areas, ponds & estuary	winter, spring & fall
Delaware	sport & commercial	handlines fixed gill net	coastal areas and Canal	winter & spring
Maryland	sport & commercial	handlines drift & fixed gill nets[d] pound net haul seine	Chesapeake Bay and rivers	fall, winter, spring & summer
Virginia	sport & commercial	handlines pound net haul seine fixed gill net fyke net	Chesapeake Bay, York, Rappahannock and James[b] Rivers	fall, winter, spring & summer
North Carolina	sport & commercial	handlines pound net otter trawl haul seine fixed gill net fyke net	Albemarle and Pamlico Sounds Roanoke River coastal areas offshore	fall, winter, spring & summer
Oregon	commercial	fixed and drift gill nets	coastal	

[a] winter = January-March, spring = April-June, summer = July-September, fall = October-December.

[b] Now closed due to public health laws regarding consumption of contaminated fish flesh.

[c] Ocean fishing prohibited within 3-mile limit.

[d] Licensed.

(Source of information for table include Koo, 1970; Chapoton and Sykes, 1961; U.S. Fishery Statistics; and personal observations by authors.)

TABLE 50. PERCENTAGES OF STRIPED BASS LANDED CAUGHT BY EACH GEAR, BY STATE, FOR 1962-66[a] and 1967-71[b]

STATE	HANDLINE		FLOATING TRAP		POUND NET		OTTER TRAWL	
	1962-66	1967-71	1962-66	1967-71	1962-66	1967-71	1962-66	1967-71
Massachusetts	93.2	93.9	0.2	0.6	6.0	4.3	-	0.05
Rhode Island	33.8	48.0	56.2	28.6	-	-	6.4	12.8
New York	2.4	9.1	-	-	3.5	5.7	14.6	8.2
New Jersey	0.6	0.8	-	-	2.2	5.6	75.9	57.6
Delaware	-	2.0	-	-	-	-	3.3	-
Maryland	3.7	2.8	-	-	6.1	7.2	0.4	6.7
Virginia	-	0.8	-	-	30.3	32.5	0.3	15.8
No. Carolina	-	-	-	-	20.3	5.2	-	14.3
ALL STATES	7.9	11.7	1.2	0.5	13.1	10.4	7.6	11.3

STATE	HAUL SEINE		FIXED GILL NET		DRIFT & RUNAROUND GILL NET		FYKE NET & OTHER	
	1962-66	1967-71	1962-66	1967-71	1962-66	1967-71	1962-66	1967-71
Massachusetts	-	-	-	0.02	-	-	0.6	0.7
Rhode Island	2.4	1.3	-	-	-	-	1.2	3.7
New York	71.6	49.4	5.3	6.6	2.6	9.5	-	-
New Jersey	3.2	2.7	9.0	12.6	6.0	12.7	3.1	8.1
Delaware	-	-	96.7	88.3	-	0.8	-	4.1
Maryland	9.0	4.5	55.3	53.3	25.4	25.5	0.1	0.1
Virginia	29.1	15.6	29.3	22.9	3.5	8.7	7.5	5.2
No. Carolina	17.3	21.9	58.9	56.3	0.7	0.2	2.8	4.2
ALL STATES	19.8	16.9	36.5	35.2	11.9	13.3	2.0	2.6

[a] 1962-66 taken from Koo, 1970

[b] 1967-71 extracted from U.S. Fish. Stat. by authors.

TABLE 51. STATE REGULATIONS GOVERNING STRIPED BASS FISHING

State	Size Limit Minimum cm	Size Limit Maximum	Fishery and Limits	Season	Sale
Maine	none	none	sport	none	no restriction
New Hampshire	40.6 FL	none	sport	none	no restriction
Massachusetts	40.6 FL	none	sport	none	no restriction
Rhode Island	40.6 FL	none	sport; commercial[a]	commercial	no restriction
Connecticut	40.6 FL	none	sport	none	prohibited[b]
New York	40.6 FL	none	sport; commercial	Hudson & Delaware Rivers closed	no restriction
New Jersey	45.7 TL	none	sport (10/day)	yes	no restriction
Delaware	30.5 TL	9.1 kg	sport; commercial	commercial	no restriction
Maryland	30.5 TL	6.8 kg	sport (1/day); commercial	none	no restriction
Virginia	35.6 TL	101.6 cm	sport (max. 2/day > 101.6 cm) commercial	none	no restriction
North Carolina	30.5 FL	none	sport; commercial	none	inland prohibited
South Carolina	25.4 TL	none	sport (2-10/day)	none	prohibited[b]
Georgia	33.1 FL	none	sport (5/day)	none	prohibited[b]
Florida	38.1 FL	none	sport (6/day)	none	prohibited[b]
Alabama	none	none	sport (5/day freshwater)	none	prohibited
Mississippi	38.1 FL	none	sport (3/day freshwater)	none	prohibited
Louisiana[c]	none	none	sport (2/day)	none	prohibited[b]
Texas	none	none	sport (landlocked)	none	prohibited[b]
California[c]	40.6 TL	none	sport (3/day)	none	prohibited[b]
Oregon	40.6 TL	none	sport (5/day); commercial	none	sport prohibited
Washington	none	none	none	none	no restriction

a Refers to states permitting use of commercial gear specifically to capture striped bass.

b Sale of striped bass taken by any gear prohibited; all other states permit sale if taken incidently in nets set for other species.

c Require salt water license; all others require freshwater fishing license if sport fishing gear is used in designated inland waters.

(Source of information for table Pfuderer et al., 1975).

Worth (1912) discussed fresh water angling grounds in North Carolina, Maryland, and Rhode Island. Other freshwater areas for angling are mentioned in Moss (1974) and Rosko (1966).

Depth Ranges --

Striped bass are pelagic and fishing depth depends greatly on the type of gear and the total depth available in the particular fishing area.

Conditions of the grounds --

Some grounds have been closed as a result of possible public health hazard from consumption of tainted fish flesh. These include the Hudson River, New York, and the James River, Virginia, specifically for PCBs and Kepone bioconcentration, respectively.

Water flow through spawning and fishing grounds may change due to diversions from hydroelectric power generation as in the Roanoke River (Fish and McCoy, 1959), or to irrigation diversion as in the Sacramento-San Joaquin Rivers (Chadwick _et_ _al_., 1977).

Fishing seasons

General pattern of season(s) --

General pattern of seasons (see Table 49) depends greatly on feeding and spawning migrations of striped bass. For example Koo (1970), tabulating landings by month by state for 1961-1966, observed 10% or more of the total catch occurred during June-October in Massachusetts, but during March-April in Maryland and Virginia, respectively.

Dates of beginning, peak and end of season(s) and variations in duration --

Table 49 shows approximate times of fishing seasons for striped bass. The season also depends on the type of gear fished, which is often directly influenced by the size of bass in the area and/or the size desired.

Ice may reduce or prevent fishing. Environmental changes responsible for changes in distributions can affect fishing seasons.

Fishing operations and results

Estimates of sport angling effort for striped bass are presented in Table 52 from the literature. In a recent creel survey of bass fishing in the Annapolis River, Nova Scotia, Jessop and Doubleday (1976) found that anglers caught an average of 0.11 bass per 2 hours of effort. The range in bass caught per hour of effort was 0-0.22 during the 1975 survey.

TABLE 52. ESTIMATES OF STRIPED BASS FISHING EFFORTS FROM SURVEY OF ANGLERS

Area	Year	% Bass of Total Caught	Catch of Bass Per Unit Specified	Author
Connecticut River	1972	4.8	-	Marcy & Galvin (1973)
Long Island	1956-1960	0-100	0-1.3/unit effort	Briggs (1962)
	1961-1963	0-100	0-0.8/unit effort	Briggs (1965)
Susquehanna River	1959-1960	0.33-1.64	-	Plosila (1961)
	1970	4.34	-	Carter (1973)
Patuxent River	1960	8.8-9.4	7-15/100 man hrs of effort	Shearer et al. (1962)
Northeast River	1958	<0.5	-	Elser (1960)
Potomac River	1959-1961	20.7-76.4	0.3-0.71/man hour	Frisbie & Ritchie (1963)
Chesapeake Bay, Virginia	1955-1960	-	0.01-0.26/man hour	Richards (1962)
Kerr Reservoir, Virginia	1961	32.95	0.02-0.11/man hour	Domrose (1963)
	1962	9.60	0.01-0.04/man hour	
Roanoke River, North Carolina	1956-1969	-	3.01-8.37/boat day	Hassler & Hogarth (1970)
Sacramento-San Joaquin Rivers	1936-1948	-	10-25 annual catch/ successful angler	Calhoun (1950)
	1938-1948	0-79	0-3.0/angler/day	Chadwick (1949)
Sacramento-San Joaquin Rivers	1951	10.8-52.0	3.5-47.6 mean catch/ successful angler	Calhoun (1953)
	1953-1959	-	0-2.3/angler/day	Chadwick (1962)
	1960-1968		0.3-2.51/angler/day	McKechnie & Miller (1971)

Commercial units of effort, landings per unit of effort, and catch per gear unit for selected states are shown in Figure 47. This is a composite from Koo (1970) of four separate figures he presented. Time series analysis (1947-71) of landings per fishing unit (Van Winkle et al., 1979b) showed an average annual increase of 17% per year for New York and 2-8% per year for Massachusetts, Maryland and North Carolina. A more detailed time series analysis (1930-1974) by these authors of Maryland landings and landings per fishing unit suggested that a portion of the long-term increase in landings was associated with gear effects rather than actual population increase. Effort and catch per unit for Maryland's haul seine fishing during 1958 and 1959 was presented by Murphy (1960). Intensity of fishing by selected gears in states where landings are legal can be seen from information in Table 50 for two, five-year periods.

Variation in fishing effort and intensity can result from entry into the stock of a dominant year-class. This occurred in Virginia when the 1966 year-class entered the gill net fishery (Grant et al., 1971). Dominant year-classes from 1934, 1940, 1958, 1966 and 1970 are represented in the landings beginning three years later.

Selectivity --

Grant and Joseph (1969) used catch statistics from pound net and fyke net fisheries in Virginia rivers as a source of non-selective (i.e., catch proportional to actual age composition) gear data. They found the haul seine and sport fisheries were selective primarily because of seasonal distribution and schooling of young bass during the summer season.

Vladykov and Wallace (1952) discussed gear versus size of bass caught. In general, they observed that pound nets took bass ranging from 10 cm to 32 kg with seasonal variability. Haul seines took various size bass and often the larger ones. Gill nets were very selective, catching bass of a size proportional to the size of mesh employed. Mansueti (1961) provided length frequency of gill net catches for different meshes by sex and age. Trent and Hassler (1968) reported mesh sizes required to catch dominant age groups in the Roanoke River. These were 4.25 and 4.75 inch stretched mesh for age III and IV males and 5.25 and 5.50 inch stretched mesh for age IV and V females.

Angling surveys have generally found that a few sport fishermen catch the majority of the bass harvested by this gear.

Evaluation of experimental nets is available in Calhoun (1945), Miller (1977) and Texas Instruments (1977b).

Catches--

Table 53 summarizes the total catches, from landings reported by state, for the United States marine areas. Where the sale of striped bass is not prohibited (Table 51) these totals include angler caught bass. Catches made purely by recreational angling have been estimated by Clark (1962), Deuel and Clark (1968), and Deuel (1973). The estimated marine

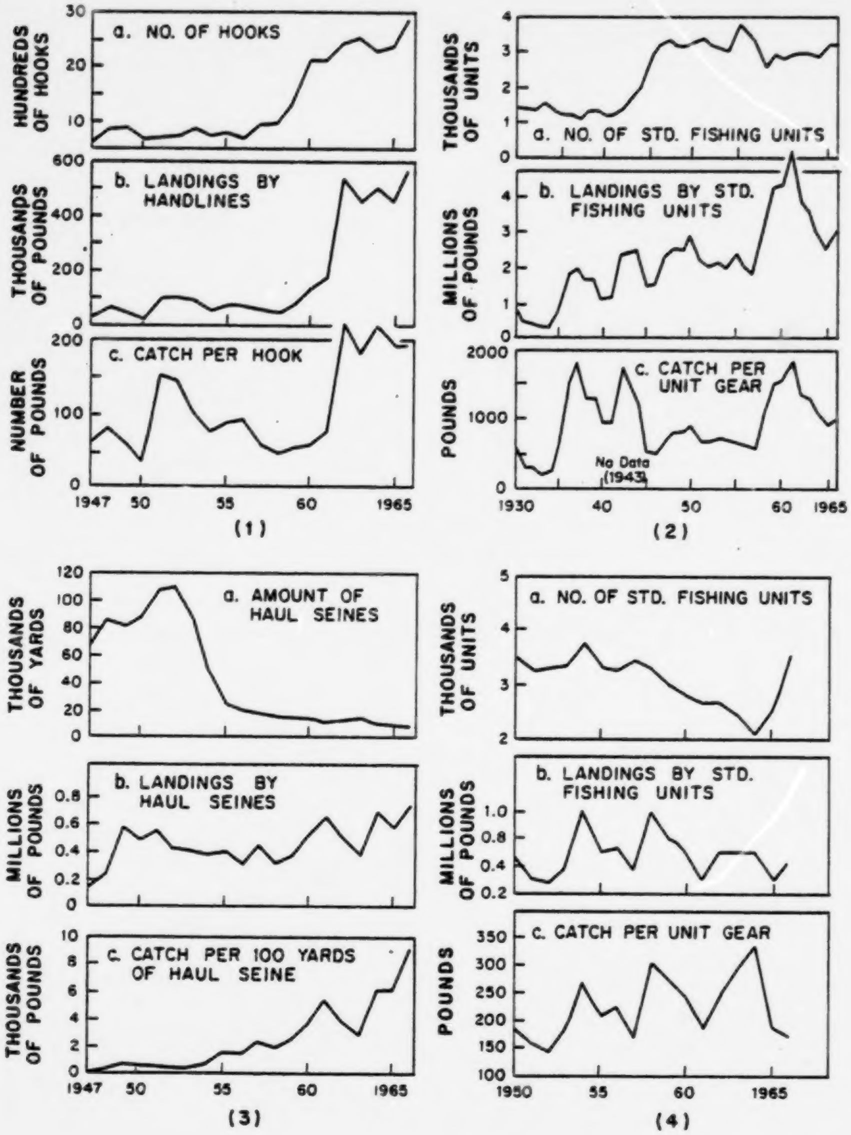


Figure 47. Landing statistics for Massachusetts (1), Maryland (2), New York (3), and North Carolina (4) striped bass fisheries. (Source: Koo, 1970, Figures 6-9)

TABLE 53. STRIPED BASS LANDINGS BY MARINE STATE IN THE UNITED STATES, IN METRIC TONS*

Year	NH	Mass	RI	Conn	NY	NJ	Del	Md	Va	NC	Ga	Calif	Ore
1930		12.3	27.2	0.9	30.0	16.8	46.3	557.5	193.0	207.5		393.6	7.3
1931		21.8	17.7	1.8	29.1	8.2	23.6	288.3	218.4	148.5		443.1	8.6
1932		14.1	3.2	1.8	14.5	5.4	3.6	197.1	269.7	230.2		271.0	8.2
1933		9.1	17.7	0.9	8.6	4.1	5.4	142.6	235.6			220.6	10.9
1934								151.2	140.7	164.3		363.7	10.9
1935		2.3	7.3	(0.5)	16.8	3.6	7.7	421.3	170.3	348.7		227.9	12.7
1936							12.3	846.3	236.1	323.7			13.2
1937		54.9	143.9	5.9	59.9	109.4	14.5	913.0	456.3	237.4			15.4
1938		37.2	95.3	4.1	63.1	66.7	11.4	778.2	524.4	153.9			20.0
1939		28.6	96.7	4.1	83.5	110.3	9.1	785.0	437.7	245.2			35.0
1940		34.5	29.1	3.6	76.7	78.1	18.6	535.7	299.2				29.1
1941								555.2	392.2				29.5
1942	3.6	44.5	43.1	8.2	120.8	43.1	26.8	1138.6	353.2				25.0
1943	8.6	45.4	33.1	11.4	143.9	72.6	16.8						30.0
1944	4.8	66.7	55.4	7.7	228.8	116.7	17.7	1217.2	846.3				43.1
1945	4.1	84.4	43.1	12.3	136.7	189.8	28.6	701.4	962.0	276.5			113.5
1946	4.1	73.1	98.5	8.6	218.8			733.2	946.1				88.1
1947	0.7	25.0	23.6	5.0	110.8	27.2	49.5	1061.5	783.2				41.8
1948		35.4	28.6	4.5	161.6	18.6	163.9	1203.1	1113.2				53.1
1949		32.7	36.8	4.1	281.2	9.5	115.8	1193.6	868.5				11.4
1950		21.3	50.8	3.2	234.7	49.5	123.0	1379.3	1269.4	361.8			16.8
1951		59.9	50.8	10.0	284.2	63.6	97.6	1060.5	819.0	318.7			12.7
1952		56.8	23.2	5.0	220.6	243.3	54.5	986.1	563.9	293.7			8.2
1953		47.7	37.2	2.7	218.8	197.5	48.1	1045.6	364.6	343.7			14.5
1954		30.9	52.7	(0.5)	199.3	23.2	66.3	957.0	431.8	509.4			10.0
1955		32.7	15.4	(0.5)	229.7	15.9	39.9	1167.7	405.9	334.1			12.3
1956		32.2	11.8	0.5	179.3	22.7	12.7	976.1	451.7	346.9	(0.5)		15.4
1957		25.4	10.4	0.5	251.1	59.9	7.3	844.0	421.8	271.0	(0.5)		5.9
1958		23.2	18.6	1.4	180.7	26.8	10.0	1409.7	597.9	497.6	0.5		10.0
1959		36.8	14.1	3.6	244.3	89.0	5.1	1974.5	952.0	395.9	(0.5)		9.1
1960		58.6	34.9	2.3	331.9	51.8	11.4	2001.7	1034.2	355.0	0.5		13.2
1961		95.3	75.8	9.1	413.1	125.3	30.0	2455.2	841.7	249.7	0.5		15.0
1962		267.4	27.7	14.5	298.3	224.3	49.0	1806.5	882.5	339.1			
1963		217.9	32.2	13.6	305.5	341.9	21.8	1702.0	1247.1	334.1	0.5		31.3
1964		237.0	34.1	15.9	415.7	452.2	14.1	1498.2	857.6	324.2	1.4		20.9
1965	3.6	210.2	27.2		336.0	345.5	14.5	1338.9	1004.7	219.7	0.9		19.1
1966	3.6	265.6	113.5		476.7	143.0	29.1	1519.5	1272.6	296.5	0.5		21.8
1967	3.6	300.5	59.9		740.0	148.5	30.0	1884.1	761.4	824.9	(0.5)		14.5
1968	4.5	396.8	44.5		704.2	208.1	22.2	2057.5	732.8	868.1	0.5		12.3
1969	5.4	171.3	59.9		696.9	143.0	19.1	2310.0	1212.6	711.9	0.5		17.7
1970	6.4	610.2	38.1		607.5	101.2	21.5	1806.0	808.6	1052.4	0.9		22.7
1971	6.8	340.0	59.5		537.5	131.7	17.7	1215.3	554.3	657.8	0.9		30.4
1972	7.3	533.0	140.3		379.5	169.3	112.6	1466.0	1207.2	572.5	2.3		25.0
1973	6.8	629.2	282.8		790.4	347.8	266.0	2259.1	1311.2	795.4	2.7		18.2

* Koo (1970) for 1930-1966 and U.S. Fishery Statistics, 1930-1973, for states not reported by Koo; all converted from thousands of pounds.

recreational catch for 1970 was 38.04 metric tons for all United States regions. For each region this total was broken down as follows:

North Atlantic (Maine-New York)	20.79 metric tons
Middle Atlantic (New Jersey-Cape Hatteras, NC)	12.38 metric tons
South Atlantic (Cape Hatteras, NC-Southern Florida)	0.09 metric tons
North Pacific (Pt. Conception, Calif.-Washington)	4.76 metric tons

(U.S. Dept. of Commerce, NMFS, 1976)

Total commercial landings for the United States were given as 5089.8 metric tons for 1974 and 3906.2 metric tons for 1975. The 1975 landings broken down by distance caught off the United States coast were given as 3685.1 metric tons for 0 to 3 miles (0 to 4 km), 212.1 metric tons for 3 to 12 miles (4 to 19 km) and 9.08 metric tons for 12 to 200 miles (19 to 322 km) by the U.S. Department of Commerce (1976).

Striped bass do not appear in great quantity in Canadian catches. Leim and Scott (1968) stated that the catch from the Canadian Atlantic for 1962 was 8.1 metric tons. They reported that about 1000 bass were angled in Shubenacadie Lake in 1949. Dadswell (1976) provided the commercial landings of striped bass by the Belleisle Bay fishery from 1895 through 1975. The smallest catch occurred in 1975 (0.68 metric tons), while the greatest were in 1966 (21.38 metric tons) and 1959 (19.80 metric tons).

The Yearbook of Fishery Statistics, FAO (1975) shows Canadian catches of striped bass (bar d'amerique) for 1974 as 10 metric tons with "more than zero but less than 50 metric tons" caught during 1970-1973. Japan reported catching 1 metric ton of striped bass in the area (21) north of Cape Hatteras in the Atlantic Ocean along the US-Canadian coast in 1974 with data not available for 1970-1973. According to this source, the United States landed 5097 metric tons in 1974 from all areas (21, 31, 67) reporting striped bass catches. Thus the total catch of striped bass for 1974 was 5118 metric tons. Unfortunately, this data is not available for earlier years so a comparison is not possible as it is for United States marine catches (Table 53).

MANAGEMENT

Regulatory (legislative) measures

Limitation of reduction of total catch --

Limitations on the fishery are summarized in Table 51 for coastal states. The states where stocks have been introduced have freshwater angling in the reservoirs and lakes.

The jurisdiction of the U.S. Fishery Conservation and Management Act of 1976 (P.L. 94-265) over this species has not been established. If this law is amended to include fisheries within the territorial limit, three Regional Councils (i.e., New England, Mid-Atlantic, and South Atlantic) along the Atlantic coast plus the Gulf and Pacific Councils would be charged with the task of developing a management plan for the striped bass fishery within their geographical areas. In the case of a migratory species such as the striped bass, the Regional Councils are provided with mechanisms to coordinate their activities and plans.

Currently, management of the inshore fisheries (including striped bass) resides with the individual states, with the State-Federal Fisheries Management Program (SFFMP) of the National Marine Fisheries Service (NMFS) coordinating the interstate programs. The SFFMP works with and often through the Atlantic States Marine Fisheries Commission (ASMFC), a compact of the fifteen Atlantic coastal states (Maine to Florida). In 1976, the Advisory Committee of the ASMFC (1977) suggested that the SFFMP undertake the job of developing a regional management plan for the striped bass fisheries within the "migratory population" from North Carolina to Maine. One result of this suggestion was the Striped Bass Management Workshop held September 23, 1977, in Baltimore, Maryland. This workshop was co-sponsored by the Northeast Region of the NMFS, SFFMP, and the Maryland Fisheries Administration. One of the recommendations of this workshop was that a striped bass management program be established through the SFFMP with funds supplied by NMFS with a full time manager* to coordinate the program. This program was organized and implemented and is administered through the established network of the ASMFC.

Pending before the 96th U.S. Congress is a bill reauthorizing the Anadromous Fish Conservation Act (U.S. Senate, 1979). If this bill and its appropriations are enacted, the NMFS (Commerce Department) and the Fish and Wildlife Service (Interior Department) will be charged under public law with conducting studies on the status of striped bass in Atlantic coastal waters as well as determining aspects of its biological and economic significance.

Protection of portions of population --

Areas and seasons of closed fishing are presented in Table 51. Size limitations as well as restrictions on use are also shown in this table.

Control or alteration of physical features of the environment

Regulation of water flow --

The completion of the Pinopolis Dam on the Cooper River to control water flow in the Santee and Cooper Rivers created the Santee-Cooper Reservoir in South Carolina. The striped bass trapped in the reservoir plus those using the navigation lock at the Pinopolis Dam have established a successfully reproducing stock which completes its entire life cycle in freshwater (Scruggs and Fuller, 1955).

---

*Michael Leverone, Project Manager, Striped Bass Management Program, Maryland Fisheries Administration, Tawes Office Bldg., Annapolis, Md. 21401.

The alteration of flow with hydroelectric plants as in the Santee-Cooper (above) and Roanoke (Fish and McCoy, 1959) Rivers, or with irrigation diversions as in the Sacramento-San Joaquin Delta (Chadwick et al., 1977), or with power plant cooling systems as in the Hudson, Potomac and other coastal rivers affects spawning sites and abundance of early life history stages.

Control of erosion and silting --

No specific attention has been given to control of silting or erosion, although patterns of siltation are affected by dam construction.

Fishways at artificial and natural obstructions --

The only fishway reportedly used by migrating striped bass is the navigation lock at the Pinopolis Dam on the Tailrace Canal of the Santee-Cooper Reservoir, South Carolina (Scruggs and Fuller, 1955).

Fish Screens --

Use of fish screens alone by bass at the Tracy Pumping Plant (Erkkila et al., 1950) and together with collectors to relocate bass at the Centra Costa Steam Plant (Kerr, 1953) has been successful in California. Experiments were also conducted with a vertical baffle type fishway for use in diversion canals (Fisk, 1959). Traveling fish screens are usually part of the current power plant construction along coastal rivers.

Control or alteration of chemical features of the environment

Water pollution control --

Chittenden (1971) stated that gross pollution had destroyed the spawning and nursery areas of striped bass in the Delaware River. Kohlhorst (1973) implicated chemical pollution in a California bass kill. Boyle (1970) warned of increasing PCB bioconcentrations in bass. The Hudson River is now closed to landings of striped bass due to the public health danger of consuming fish high in PCBs. Kepone pollution has similarly closed the James River. Mansueti (1962) discussed increasing pollution and striped bass survival. Kumpf (1977) reviewed the economic impact on sport and commercial fisheries, including striped bass, of possible pollution resulting from oil and gas exploration and production, effluent discharge resulting in fish kills, and habitat alteration by dredging and filling. The coastal areas of his impact assessment were the North Atlantic, Mid-Atlantic, South Atlantic, and Gulf of Mexico regions.

Radtke and Turner (1967) investigated apparent blockage of spawning migration by high total dissolved solids in the San Joaquin River. They concluded that some planned water diversions could threaten entire spawning runs.

Artificial fertilization of waters

Pond management (see Section 4) is the only case of controlled fertilization. Possible effects of fertilization from pollution are discussed by Mansueti (1962) and Talbot (1966).

Control or alteration of the biological features of the environment

Control of aquatic vegetation --

This occurs primarily in management of pond culture situations of juvenile bass (Bonn *et al*., 1976). Copper sulfate has been recommended for algal control in these ponds. Casoron and Simazine have been recommended for the control of aquatic vegetation. However, the median tolerance limit of juvenile bass to Simazine (see Table 39) is much lower than the rate recommended for control.

Control of parasites and diseases --

Little parasite or disease control has been attempted except under culture conditions. These are described in Section 10 for small scale or semi-closed rearing systems. In pond management situations Lindane and Malathion have been suggested for the control of parasitic copepods. Ethyl parathion has been used in the control of predators in ponds prior to the introduction of the juvenile bass. Bayluscide has been used to control snails that are suspected to be acting as hosts for trematode parasites.

Control of predation and competition --

Controls of this nature have not been attempted, although bass are used to control some shad populations in freshwater reservoirs and lakes.

Population manipulation --

Population manipulation beyond the modeling stage has not been attempted. For a general description of these models, see Section 13 .

Artificial stocking

Maintenance stocking --

Most of the bass produced currently in state and federal hatcheries are to maintain stocks introduced to control shad populations and/or to provide sport fishing in inland waters.

Transplantation, introduction --

Striped bass were transplanted to the Sacramento River, California, in 1879 and 1882 in two shipments (via railroad) of bass from the Navesink and Shrewsbury Rivers, New Jersey. These transplanted bass (young to subadults) apparently adapted well and by 1887 extended from San Diego to

Oregon (Smith, 1896). By 1889 the commercial production in San Francisco Bay was about 1 million pounds annually (Scofield, 1931), indicating successful reproduction and growth.

With the discovery that the striped bass could complete its life cycle in freshwater (Scruggs and Fuller, 1955), inland states became interested in stocking it to control shad populations and to provide additional sport fishing (Bailey, 1975). Fingerling stocking has been reported to be more effective than the stocking of either fry or adults. Bailey (1975) provided an evaluation of stocking programs in the southeastern United States. For the current distribution of striped bass see Section 6 .

SECTION 15

REFERENCES

Al-Ahmad, T. A. 1978. Evaluation of rotifers as food for striped bass, *Morone saxatilis* (Walbaum), fry under controlled conditions. M.S. Thesis. Auburn University, Auburn, Alabama. 74 pp.

Albrecht, A. B. 1964. Some observations on factors associated with survival of striped bass eggs and larvae. Calif. Fish & Game 50(2):100-113.

Alexander, J. E., J. Foerenbach, S. Fisher, and D. Sullivan. 1973. Mercury in striped bass and bluefish. N.Y. Fish & Game Jour. 20(2):147-151.

Allen, K. O. 1972. Myxobacteria infections of striped bass. Auburn University Dept. Fish. and Allied Aquaculture, S.E. Coop Disease Project Newsletter No. 9: 3.

Alperin, I. M. 1965. Recent records of pugheaded striped bass from New York. N.Y. Fish & Game Jour. 12(1):114-115.

Alperin, I. M. 1966a. Dispersal, migration and origins of striped bass from Great South Bay, Long Island. N.Y. Fish & Game Jour. 13(1):79-112.

Alerpin, I. M. 1966b. Occurrence of yearling striped bass along the south shore of Long Island. N.Y. Fish & Game Jour. 13(1):113-120.

Amlacher, E. 1970. Textbook of fish diseases. Translated by D.A. Conroy and R.L. Herman. T.F.H. Publications, Inc., New Jersey. 302 pp.

Anderson, J. C. 1966. Production of striped bass fingerlings. Progr. Fish-Cult. 28(3):162-164.

APHA (American Public Health Association). 1965. Standard methods for the examination of water and wastewater including bottom sediments and sludges. 12th Edition. American Public Health Association, Inc. New York. 769 pp.

Atlantic States Marine Fisheries Commission. 1977. Thirty-sixth annual report on the ASMFC to the Congress of the United States. 42 pp.

Auld, A. H., and J. R. Schubel. 1978. Effects of suspended sediments on fish eggs and larvae: A laboratory assessment. Estuarine and Coastal Marine Sciences 6:153-164.

Austin, H. M., and O. Custer. 1977. Seasonal migration of striped bass in Long Island Sound. N.Y. Fish and Game Jour. 24(1):53-68.

Austin, H. M., and C. R. Hickey, Jr. 1978. Predicting abundance of striped bass, *Morone saxatilis*, in New York waters from modal lengths. U.S. Fish. Bull. 76(2):467-473.

Bailey, W. M. 1975. An evaluation of striped bass introductions in the southeastern United States. Proc. 28th Ann. Conf. Southeastern Game and Fish Comm. pp. 54-68.

Barkuloo, J. M. 1970. Taxonomic status and reproduction of striped bass *(Morone saxatilis)* in Florida. U.S. Fish & Wildl. Serv., Bur. Sport Fish. & Wildl. Tech. Papers 44:1-16.

Barnthouse, L. W., et al. (Energy Division). 1977. A selective analysis of power plant operation on the Hudson River with emphasis on the Bowling Point Generating Station. Oak Ridge National Laboratory, Oak Ridge, Tennessee, Contract No. W-405-eng-26. ORNL/TM-5877/V1 and 2.

Bason, W. H. 1971. Ecology and early life history of striped bass, *Morone saxatilis*, in the Delaware estuary. Ichthological Associates, Bull. 4:1-122.

Bason, W. H., S. E. Allison, L. O. Horseman, W. H. Keirsey and C. A. Shirey. 1975. Fishes. *In* Ecological Studies in the Vicinity of the Proposed Summit Power Station. January through December 1974. Vol. I. Ichthyological Associates, Inc. for Delmarva Power and Light Co., July 1975. 327 pp.

Bayless, J. D. 1968. Striped bass hatching and hybridization experiments. Proc. 21st Ann. Conf. Southeastern Assoc. of Game and Fish Comm.: 232-244.

Bayless, J. D. 1972. Artificial propagation and hybridization of striped bass, *Morone saxatilis* (Walbaum). S.C. Wildlife and Marine Resources Dept. 135 pp.

Beck, A. D., D. A. Bengtson and W. H. Howell. 1980. International Study on *Artemia*. V. Nutritional value of five geographical strains of *Artemia*: effects on survival and growth of larval Atlantic silversides, *Menidia menidia*. *In* The brine shrimp *Artemia*. Vol. 3. Ecology, Culturing, Use in Aquaculture (G. Persoone, P. Sorgeloos, O. Roels and E. Jaspers, eds.). Universa Press, Wetteren, Belgium. pp.

Beck, A. P., G. V. Poje, and W. T. Waller. 1975. A laboratory study on the effects of the exposure of some entrainable Hudson River biota on the hydrostatic pressure regimes calculated for Storm King. *In* Fisheries and Energy Production, A Symposium (Saul B. Saila, ed.). D.C. Heath Publishing Co., Lexington, Mass. pp. 167-204.

Bengtson, D. A., A. D. Beck, and H. A. Poston. 1978. Comparative effects of live and artificial diets on growth and survival of juvenile Atlantic silverside, *Menidia menidia*. Proceeding of the World Mariculture Society 9:159-173.

Benville, P. E., and S. Korn. 1977. The acute toxicity of six monocyclic aromatic crude oil components to striped bass (*Morone saxatilis*) and bay shrimp (*Crago franciscorum*). Calif. Fish & Game 63(4):204-209.

Berg, L. S. 1949. Freshwater fishes of the U.S.S.R. and adjoining countries. (Tabl. Anal. Fauna URSS, vol. 30.) Acad. Sci. USSR Moscow, 3:929-1381, figs. 675-945. (In Russian.)

Berggren, T. J., and J. T. Lieberman. 1978. Relative contribution of Hudson, Chesapeake, and Roanoke striped bass, *Morone saxatilis*, stocks to the Atlantic coast fishery. U.S. Fish. Bull. 76(2):335-345.

Bibko, P. N., L. Wirtenan, and P. E. Kuser. 1974. Preliminary studies on the effects of air bubbles and intense illumination on the swimming behavior of the striped bass (*Morone saxatilis*) and the gizard shad (*Dorosonia cepedianum*). *In* Entrainment and Intake Screening. Proc. 2nd Workshop on Entrainment and Intake Screening (L. D. Jensen, ed.), The Johns Hopkins Univ., Baltimore, Maryland. pp. 293-304.

Bigelow, H. B., and W. C. Schroeder. 1953. Striped bass. *In* Fishes of the Gulf of Maine. U.S. Fish & Wildl. Serv., Fish Bull. 54(53):389-405. Revision of U.S. Bur. Fish. Bull. No. 40.

Bischoff, A. E. 1970. Pesticides Investigation. Current Fish and Wildlife Problems. Calif. Dept. of Fish and Game, FW-001-R-08. 3 pp.

Bishop, R. D. 1968. Evaluation of the striped bass (*Roccus saxatilis*) and white bass (*R. chrysops*) hybrids after two years. Proc. 21st Ann. Conf. Southeastern Assoc. Game and Fish Comm.: 245-254.

Bishop, R. D. 1975. The use of circular tanks for spawning striped bass (*Morone saxatilis*). Proc. 28th Ann. Conf Southeastern Assoc. Game and Fish Comm.: 35-44.

Blaxhall, P. C. 1972. The haematological assessment of the health of freshwater fish. A review of selected literature. J. Fish. Biol. 4:593-604.

Bloch, M. E. 1785-97. Ichthylogie, on Histoire naturelle, generale et particuliere des Poissons. 12v. Fr. de la Garde, Berlin.

Blondin, G. A., B. D. Kulkarni, and W. R. Nes. 1967. A study of the origin of vitamin-D from 7-dehydrocholesterol in fish. Comp. Biochem. Physiol. 20(2):379-390.

Blondin, G. A., J. L. Scott, J. K. Hummer, B. D. Kulkarni, and W. R. Nes. 1966. The biosynthesis of squalene and sterols in fish. Comp. Biochem. Physiol. 17(2):391-407.

Bogdanov, A. S., S. I. Doroshev, and A. F. Karpevich. 1967. Experimental transfer of *Salmo gairdneri* (Richardson) and *Roccus saxatilis* (Walbaum) from the USA for acclimatization in waters of the USSR. Voprosy Ikhtiologii, Akademiya Nauk SSSR 7(1):185-187. Translated from Russian by Robert M. Howland, fishery biologist, Narragansett Marine Game Fish Research Laboratory, Bur. Sport Fish. and Wildl.

Bonn, E. W. 1970. Striped bass introduction. Tex. Parks and Wildlife Dept. Job Competion Rept., F-08-R-17. 12 pp.

Bonn, E. W., W. M. Bailey, J. D. Bayless, K. E. Erickson, and R. E. Stevens (eds.). 1976. Guidelines for striped bass culture. Striped Bass Committee, Southern Division, American Fisheries Society. 103 pp.

Bonner, R. R., Jr. 1965. Observations on tag loss and comparative mortality in striped bass. Ches. Sci.,6(3):197-198.

Boone, J. 1973. Pesticides, PCB's, and striped bass reproduction. Maryland Fish and Wildlife News, 3(12):1-3.

Bowen, J. T. 1970. A history of fish culture as related to the development of fishery programs. *In* A Century of Fisheries in North America (N. G. Benson, ed.). American Fisheries Society Special Publication No. 7:71-93.

Bowles, R. R. 1976. Effects of water velocity on activity patterns of juvenile striped bass. Proc. Ann. Conf. Southeastern Assoc. Game Fish Comm., 29:142-151.

Bowker, R. G., D. J. Baumgartner, J. A. Hutcheson, R. H. Ray, and T. L. Wellborn, Jr. 1969. Striped bass *Morone saxatilis* (Walbaum) 1968 report on the development of essential requirements for production. U.S. Fish and Wildl. Serv. Publ. 111 pp.

Boyle, R. H. 1970. Poison roams our coastal seas. Sports Illustrated, 33(17):70-74, 76, 81, 84 (October 26, 1970).

Braid, M. R. 1977. Factors affecting the survival and growth of striped bass, *Morone saxatilis* (Walbaum), fry in recirculating systems. Ph.D. Thesis, Auburn University, Auburn, Alabama. 86 pp. (University Microfilms, Ann Arbor, Michigan, Order No. 77-24,492.)

Braschler, E. W. 1975. Development of pond culture techniques for striped bass. Proc. 28th Ann. Southeastern Game and Fish Comm.: 44-48.

Brauhn, J. L., and R. A. Schoettger. 1975. Acquisition and culture of research fish: rainbow trout, fathead minnows, channel catfish, and bluegills. EPA-660/3-75-011. U.S. Environmental Protection Agency, Corvallis, Oregon. 52 pp.

Brice, J. J. (Commissioner). 1898. A manual of fish-culture, based on the methods of the U.S. Commission of Fish and Fisheries, with chapters on the cultivation of oysters and frogs. Rept. U.S. Fish Comm. for 1897, Appendix: 1-340. Doc. No. 299.

Briggs, P. T. 1962. The sport fisheries of Great South Bay and vicinity. N.Y. Fish & Game Jour., 9(1):1-36.

Briggs, P. T. 1965. The sport fishery in the surf on the south shore of Long Island from Jones Inlet to Shinnecock Inlet. N.Y. Fish & Game Jour., 12(1)31-47.

Brockson, R. W., and H. T. Bailey. 1974. Respiratory response of juvenile chinook salmon and striped bass exposed to benzene, a water-soluble component of crude oil. Proceedings of 1973 Conference on Prevention and Control of Oil Pollution. Washington, D.C., p. 783-790.

Brothers, E. B., C. P. Mathews, and R. Lasker. 1976. Daily growth increments in otoliths from larval and adult fishes. U.S. Fish Bull., 74(1): 1-8.

Brown, B. E. 1965. Meristic counts of striped bass from Alabama. Trans. Amer. Fish. Soc., 94(3):278-279.

Bryant, C. P., and H. R. Seibel. 1971. Ultrastructural investigation of tubulogenesis in the mesonephros of the striped bass, Roccus saxatilis. Anat. Rec., 169(2):285.

Bulak, J. S. 1976. Investigations into the intial inflation of the swimbladder in striped bass (Morone saxatilis). M.A. Thesis, Southern Illinois University, Carbondale, Illinois. 65 pp.

Burton, D. T., L. W. Hall, Jr., S. L. Margery, and R. D. Small. 1979. Interactions of chlorine, temperature change (ΔT), and exposure time on survival of striped bass (Morone saxatilis) eggs and prolarvae. J. Fish. Res. Board Can., 36(9):1108-1113.

Calhoun, A. J. 1946. Observations of the striped bass fishery in the Sacramento Delta area during April and May of 1946. Rept. Bur. of Fish Cons., Calif. Div. of Fish & Game, Inland Fish. Br., Admin. Rept. No. 46-12. 36 pp.

Calhoun, A. J. 1950. California angling catch records from postal card surveys: 1936-1948; with an evaluation of postal card non-response. Calif. Fish & Game, 36:178-234.

Calhoun, A. J. 1952. Annual migration of California striped bass. Calif. Fish & Game, 28:291-403.

Calhoun, A. J. 1953a. Aquarium tests of tags on striped bass. Calif. Fish & Game, 39:209-218.

Calhoun, A. J. 1953b. State-wide California angling estimates for 1951. Calif. Fish & Game, 39(1):103-113.

California Department of Fish and Game. 1974. Interagency Ecological Study Program for the Sacramento-San Joaquin Estuary. A cooperative study by the Calif. Dept. of Fish & Game, Calif. Dept. of Water Resources, U.S. Bureau of Sport Fisheries and Wildlife, and the U.S. Bureau of Reclamation. Third Annual Report (1973). 81 pp.

Carlson, F. T., and J. A. McCann. 1969. Report on the biological findings of the Hudson River fisheries investigations, 1965-1968. *In* Hudson River Fisheries Investigations, 1965-1968: Evaluations of a proposed pump storage project at Cornwall, New York, in relation to fish in the Hudson River. Hudson River Policy Committee, New York State Conservation Dept. 50 pp. & appendix.

Carpenter, J. H. and V. E. Grant. 1967. Concentration and state of cerium in coastal waters. J. Mar. Res., 25(3): 228-238.

Carreon, J. A. 1978. Studies on the culture of larval striped bass, *Morone saxatilis* (Walbaum), in closed recirculating systems. Ph.D. Thesis, Auburn University, Auburn, Alabama. 112 pp. (University Microfilms, Ann Arbor, Michigan, Order No. 78-14078.)

Carter, W. R. III. 1973. Ecological study of Susquehanna River and tributaries below the Conowingo Dam, January 1, 1967 to March 31, 1971. AFSC-1 Job Completion Rept. 122 pp.

Carter, H. H., R. E. Wilson, and G. Carroll. 1979. An assessment of the thermal effects on striped bass larvae entrained in the heated discharge of the Indian Point Generating Facilities Units 2 & 3. Special Report 24 (Ref. 79-7), Marine Sciences Research Center, SUNY. 33 pp.

Catchings, E. D. 1973. The effects of increased salinity and of feeding treatments on the survival and growth of striped bass, *Morone saxatilis* (Walbaum), fry in hatching jars, and fingerling striped bass in troughs. M.S. Thesis, Auburn University, Auburn, Alabama. 61 pp.

Chadwick, H. K. 1958. A study of the planktonic fish eggs and larvae of the Sacramento-San Joaquin Delta with special reference to the striped bass (*Roccus saxatilis*). Calif. Dept. Fish & Game, Inland Fish. Br., Admin. Rept. No. 58-5. 24 pp.

Chadwick, H. K. 1960. Toxicity of Tricon Oil Spill Eradicator to striped bass (*Roccus saxatilis*). Calif. Fish & Game, 46:371-372.

Chadwick, H. K. 1962. Catch records from the striped bass sport fishery in California. Calif. Fish & Game, 48(3):153-177.

Chadwick, H.K. 1963. An evaluation of five tag types used in a striped bass mortality rate and migration study. Calif. Fish & Game, 49(2):64-83.

Chadwick, H. K. 1964. Annual abundance of young striped bass, Roccus saxatilis, in the Sacramento-San Joaquin Delta, California. Calif. Fish & Game, 50(2):69-99.

Chadwick, H. K. 1965. Determination of sexual maturity in female striped bass (Roccus saxatilis). Calif. Fish & Game, 51(3):202-206.

Chadwick, H. K. 1966. Variation in the growth of young striped bass (Roccus saxatilis) in the Sacramento-San Joaquin system. Calif. Fish & Game, Inland Fish Br., Admin. Rept. No. 66-11. 6 pp.

Chadwick, H. K. 1967. Recent migrations of the Sacramento-San Joaquin River striped bass populations. Trans. Amer. Fish. Soc., 96(3):327-342.

Chadwick, H. K. 1968. Mortality rates in the California striped bass population. Calif. Fish & Game, 54(4):228-246.

Chadwick, H. K. 1969. An evaluation of striped bass angling regulations based on an equilibrium yield model. Calif. Fish & Game, 55(1):12-19.

Chadwick, H. K., D. E. Stevens, and L. W. Miller. 1977. Some factors regulating the striped bass population in the Sacramento-San Joaquin Estuary, California. In Proceedings of the Conference on Assessing the Effects of Power-Plant-Induced Mortality on Fish Populations (W. Van Winkle, ed.). Pergamon Press, New York. pp. 18-35.

Chapotan, R. B., and J. E. Sykes. 1961. Atlantic coast migration of large striped bass as evidenced by fisheries and tagging. Trans. Amer. Fish. Soc., 90(1):13-20.

Cheek, R. D. 1966. Pugheaded striper. Wildlife in North Carolina, 30(6): 27.

Chittenden, M. E., Jr. 1971. Status of the striped bass, Morone saxatilis, in the Delaware River. Ches. Sci., 12(3):131-136.

Chittenden, M. E., Jr. 1972. Effects of handling and salinity on oxygen requirements of the striped bass, Morone saxatilis. J. Fish. Res. Bd. Canada, 28(12):1823-1830.

Christensen, S. W., D. L. DeAngelis, and A. G. Clark. 1977. Development of a stock-progeny model for assessing power plant effects on fish populations. In Proceedings of the Conference on Assessing the Effects of Power-Plant-Induced Mortality on Fish Populations (W. Van Winkle, ed.). Pergamon Press, New York. pp. 196-225

Clark, G. H. 1934. Tagging of striped bass. Calif. Fish & Game, 20(1): 14-19.

Clark, G. H. 1938. Weight and age determination of striped bass. Calif. Fish & Game, 24(2):176-177.

Clark, J. R. 1962. The 1960 salt-water angling survey, U.S. Dept. Interior, Bur. Sport Fish. Wildl., Circular 153. 36 pp.

Clark, J. R. 1968. Seasonal movements of striped bass contingents of Long Island Sound and the New York Bight. Trans. Amer. Fish. Soc., 97(4): 320-343.

Clarke, W. C. 1973. Sodium-retaining bioassay of prolactin in the intact teleost *Tilapia mossambica* acclimated to sea water. Gen. Comp. Endocrin., 21(3):498-512.

Conte, M. H., R. G. Otto, and P. E. Miller. 1979. Short-term variability in surface catches of ichthyoplankton in the Upper Chesapeake Bay. Estuarine and Coastal Marine Science, 8:511-522.

Cordonnier, L. M., and H. L. Ward. 1967. *Pomphosrhynchus rocci* sp. n. (Acanthocephala) from the rock bass, *Roccus saxatilis*. J. Parasitol., 53(6):1295-1297.

Courtois, L. A. 1974. Physiological responses of striped bass, *Roccus saxatilis* (Walbaum) to changes in diet, salinity, temperature, and acute copper exposure. Ph.D. Thesis, University of California at Davis. 130 pp. (University Microfilms, Ann Arbor, Michigan, Order No. 74-29, 297.)

Coutant, C. C., and R. J. Kedl. 1975. Survival of larval striped bass exposed to fluid-induced and thermal stresses in a simulated condenser tube. Oak Ridge National Laboratory, Contract No. W-7405-eng-26. Environmental Sciences Division, Publ. No. 637. 37 pp. (ORNL-TM-4695)

Dadswell, M. J. 1976. Notes on the biology and resource potential of striped bass in the Saint John Estuary. *In* Baseline survey and living resource potential study of the Saint John River Estuary, Vol. 3, Fish and Fisheries. The Huntsman Marine Laboratory, St. Andrews, New Brunswick.

Daniel, D. A. 1976. A laboratory study to define the relationship between survival of young striped bass (*Morone saxatilis*) and their food supply. Calif. Fish & Game, Anadromous Fish Br., Admin. Rept. No. 76-1. 13 pp.

Davies, W. D. 1970. The effects of temperature, pH and total dissolved solids on the survival of immature striped bass, *Morone saxatilis* (Walbaum). Ph.D. Thesis, North Carolina State University, Raleigh, NC. 100 pp.

Davies W. D. 1973. The effects of total dissolved solids, temperature, and pH on the survival of immature striped bass: A response surface experiment. Prog. Fish-Cult., 35(3):157-160.

Davis, H S. 1967. Culture and Diseases of Game Fishes. University of California Press, Berkeley and Los Angeles. 332 pp.

Davis, R. H., Jr. 1966. Population studies of striped bass, Roccus saxatilis (Walbaum), in Maine based on age distribution and growth rate. M.S. Thesis, University of Maine, Orono. 51 pp.

Davis, W. S. 1959. Field tests of Petersen, streamer, and spaghetti tags on striped bass, Roccus saxatilis (Walbaum). Trans. Amer. Fish. Soc., 88(4):319-329.

Dawson, M. A., E. Gould, F. P. Thurberg, and A. Calabrese. 1977. Physiological response of juvenile striped bass, Morone saxatilis, to low levels of cadmium and mercury. Ches. Sci., 18(4):353-359.

Degens, E. T., W. G. Deuser, and R. L. Haedrich. 1969. Molecular structure and composition of fish otoliths. Marine Biol., 2:105-113.

Delor, A. 1973. Elevage du bar americain Roccus saxatilis (Walbaum, 1792). I. Methodes utilisees. Sci. Peche, Bull, Inst. Peches marit. No. 224:1-19.

Dendy, J. S. 1979. Polyps of Craspedacusta sowerbyi as predators on young striped bass. Prog. Fish-Cult., 40:5-6.

Dergaleva, Zh. T., and M. I. Shatunovskiy. 1977. Data on the lipid metabolism of the larvae and young of the striped bass, Morone saxatilis. J. Ichthyl., 17(5):802-804.

DeSylva, D. P. 1962. Racial status of juvenile striped bass in the Delaware River estuary. University of Delaware, Dept. of Biological Science, Newark, Ref. No. 61-10:1-35.

DeSylva, D. P., F. A. Kalber, and C. N. Schuster. 1962. Fishes and ecological conditions in the shore zone of the Delaware River Estuary, with notes on other species collected in the deeper water. Univ. of Delaware, Marine Lab. Infor. Ser. Publ. 5. 164 pp.

Deuel, D. G. 1973. 1970 salt-water angling survey. NOAA, NMFS, Current Fishery Statistics No. 6200. 64 pp.

Deuel, D. G., and J. R. Clark. 1968. The 1965 salt-water angling survey. Bur. Sport Fish. & Wildl., Res. Publ. 67. 51 pp.

Domrose, R. J. 1963. Striped bass study. Virginia D-J Project Rept., Comm. of Game and Inland Fisheries. Completion Rept., F-5-R-8. 54 pp.

Dorfman, D., and J. Westman. 1970. Responses of some anadromous fishes to varied oxygen concentration and increased temperatures. Rutgers Univ., Water Resources Res. Inst., Partial completion and termination report. 75 pp. (PB192312)

Doroshev, S. I. 1970. Biological features of the eggs, larvae and young of the striped bass (Roccus saxatilis (Walbaum)) in connection with the problem of its acclimation in the U.S.S.R. J. Ichth., 10(2):235-278.

Doroshev, S. I., and J. W. Cornacchia. 1979. Initial swim bladder inflation in the larvae of Tilapia mossambica (Peters) and Morone saxatilis (Walbaum). Aquaculture, 16 (1979):57-66.

Dovel, W. L. 1971. Fish eggs and larvae of the upper Chesapeake Bay. Natural Resources Institute Spec. Rept. No. 4, Contrib. No. 460, Natural Res. Inst., Univ. of Maryland. 71 pp.

Dovel, W. L., and J. R. Edmunds. 1971. Recent changes in striped bass (Morone saxatilis) spawning sties and commercial fishing areas in Upper Chesapeake Bay: Possible influencing factors. Ches. Sci., 12(1): 33-39.

Dudley, R. G., A. W. Mullis, and J. W. Terrell. 1977. Movements of adult striped bass (Morone saxatilis) in the Savannah River, Georgia. Trans. Am. Fish. Soc., 106(4):314-322.

Durbin, E. G., and A. G. Durbin. 1978. Length and weight relationships of Acartia clausi from Narragansett Bay, Rhode Island. Limnol. Oceanogr., 23(5):958-969.

Ecological Analysts, Inc., and The Johns Hopkins University. 1974. Power Plant Site Evaluation, Aquatic Biology, Final Report. Douglas Point Site, Maryland, PPSE 4-2, Vol. 2. 479 pp.

Edwards, G. B. 1974. Biology of the striped bass, Morone saxatilis (Walbaum), in the lower Colorado River (Arizona-California-Nevada). M.S. Thesis, Arizona State University, Tempe. 45 pp.

Edwards, S. R., and F. M. Nahhas. 1968. Some ectoparasites of fishes from the Sacramento-San Joaquin Delta, California. Calif. Fish & Game, 54(4):247-256.

Ehrlich, K. F., J. H. S. Blaxter, and R. Pemberton. 1976. Morphological and histological changes during the growth and starvation of herring and plaice larvae. Mar. Biol., 35:105-118.

Eisler, R., G. R. Gardner, R. J. Hennekey, G. LaRoche, D. F. Walsh, and P. O. Yevich. 1972. Acute toxicology of sodium nitrilotriacetic acid (NTA) and NTA-containing detergents to marine organisms. Water Research, 6:1009-1027.

Eldridge, M. B., J. J. King, D. Eng, and M. T. Bowers. 1977. Role of the oil glouble in survival and growth of striped bass (Morone saxatilis) larvae. Proceedings 57th Annual Conference West. Assoc. State Game & Fish Comm.: 303-313.

Eldridge, M. B., J. A. Whipple, D. Eng, M. J. Bowers, and B. M. Jarvis. 1981. Effects of food and feeding factors on laboratory-reared striped bass larvae. Trans. Amer. Fish. Soc., 110(1):111-120.

Elser, H. J. 1960. Creel census results on the Northeast River, Maryland, 1958. Ches. Sci., 1(1):41-47.

Engel, D. W., and E. M. Davis. 1964. Relationship between activity and blood composition in certain marine teleosts. Copeia, 1964(3):586-587.

Eraslan, A. H., W. Van Winkle, R. D. Sharp, S. W. Christensen, C. P. Goodyear, R. M. Rish, and W. Fulkerson. 1976. A computer simulation model for the striped bass young-of-the-year population in the Hudson River. Environmental Sciences Division Publ. No. 766, Oak Ridge National Laboratory (ORNL/NUREG-8 Special). 208 pp.

Erickson, K. E., J. Harper, G. C. Mensinger, and D. Hicks. 1972. Status and artificial reproduction of striped bass from Keystone Reservoir, Oklahoma. Proc. of 25th Ann. Conf. Southeastern Assoc. of Game & Fish Comm.:513-522.

Erkkila, L. F., J. W. Moffett, O. B. Cope, B. R. Smith, and R. S. Neilson. 1950. Sacramento-San Joaquin Delta Fishery Resources: Effects of Tracy pumping plant and Delta Cross Channel. U.S. Fish & Wildl. Serv., Spec. Sci. Rept.-Fish., 56:1-109.

FAO. 1975. Food and Agriculture Organization of the United Nations, Yearbook of Fishery Statistics. Vol. 40.

Farley, T. C. 1966. Striped bass (Roccus saxatilis), spawning in the Sacramento-San Joaquin River system during 1963 and 1964. In Ecological studies of the Sacramento-San Joaquin estuary, Part II. Calif. Fish & Game Dept., Fish Bull No. 136:28-43.

*
Fish, F. F., and E. G. McCoy. 1959. The river discharges required for effective spawning by striped bass in the rapids of the Roanoke River of North Carolina. North Carolina Wildlife Res. Comm., Raleigh, N.C. 39 pp.

Fisk, L. O. 1959. Experiments with a vertical baffle fishway. Calif. Fish & Game, 45:111-122

Forrester, C. R., A. E. Peden, and R. M. Wilson. 1972. First records of the striped bass, Morone saxatilis, in British Columbia waters. J. Fish. Res. Bd. Canada, 29(3):337-339.

Freadman, M. A. 1978. Swimming energetics of striped bass and bluefish. Ph.D. Thesis, University of Massachusetts, Amherst. 97 pp. (University Microfilms, Ann Arbor, Michigan, Order No. 79-02002.)

Freeman, B. L. 1977. Notes on striped bass migrations. Underwater Naturalist, 10(4):13-19.

Freihoffer, W. C. 1960. Neurological evidence for the relationships of some percomorph fishes. Ph.D. Thesis, Stanford University. 204 pp.

Frisbie, C. M. 1967. Age and growth of the striped bass, Roccus saxatilis (Walbaum), in Massachusetts coastal waters. M.S. Thesis, University of Massachusetts, Amherst. 58 pp.

Frisbie, C. M., and D. E. Ritchie, Jr. 1963. Sport fishing survey of the lower Potomac Estuary, 1959-1961. Ches. Sci., 4(4):175-191.

Gaines, J. L., Jr., and W. A. Rogers. 1972. Fish mortalities associated with Goezia sp. (Nematoda: Ascaroidea) in Central Florida. Proc. 25th Ann. Conf. Southeastern Assoc. of Game & Fish Comm.:496-497.

Gift, J. J., and J. R. Westman. 1971. Responses of some estuarine fishes to increasing thermal gradients. Privately published monograph, June, 1971. 154 pp.

Gomez, R. 1970. Food habits of young-of-the-year striped bass, Roccus saxatilis (Walbaum) in Canton Reservoir. Proc. Okla. Acad. Sci., 50: 79-83.

Goodson, Lee F. 1964. Diet of striped bass at Millerton Lake, California. Calif. Fish & Game, 50(4):307.

Goodyear, C. P. 1978. Management problems of migratory stocks of striped bass. In Marine Recreational Fisheries (H. Clepper, ed.). Sport Fishing Institute, Washington, D.C., pp. 75-84.

Grant, G. C. 1974. The age composition of striped bass catches in Virginia rivers, 1967-1971, and a description of the fishery. U.S. Fish. Bull., 72(1):193-199.

Grant, G. C., V. G. Burrell, Jr., and W. H. Kriete, Jr. 1971. Age composition and magnitude of striped bass winter gill-net catches in the Rappahannock River, 1967-1970. Proc. of the 24th Ann. Conf. Southeastern Assoc. of Game & Fish Comm.:659-667.

Grant, G. C., and E. B. Joseph. 1969. Comparative strength of the 1966 year class of striped bass, Roccus saxatilis (Walbaum), in three Virginia rivers. Proc. 22nd Ann. Conf. Southeastern Assoc. Game & Fish Comm.:501-509.

Grant, G. C., and J. V. Merriner. 1971. Feasibility of increasing striped bass populations by stocking of underutilized nursery grounds. Anadromous Fish Project. Virginia-AFS-6-1. 89 pp. & appendices.

Green, S. 1882. Hatching striped bass, sturgeon and trout. Trans. Amer. Fish-Cult. Assoc.:37-40.

Gregory, W. K. 1918. The structure and mechanism of fishes. In Fishes of the vicinity of New York City (John T. Nichols, ed.). Amer. Mus. Nat. Hist., Handb. Ser. 7:5-17, fig. 7, 1 pl.

Gregory, W. K. 1933. Fish skulls: a study of the evolution of natural mechanisms. Trans. Amer. Philos. Soc., n.s. 23, pt. 2: vii-75-481, fig. 302.

Grinstead, R. G. 1971. Effects of pugheadedness on growth and survival of striped bass, *Morone saxatilis* (Walbaum), introduced into Canton Reservoir, Oklahoma. Proc. Okla. Acad. Sci., 51:8-12.

Grove, T. L., T. J. Berggren, and D. Powers. 1976. The use of innate tags to segregate spawning stocks of striped bass (*Morone saxatilis*). Estuarine Processes, Vol. 1 (Wiley, ed.). Academic Press, N.Y. pp. 166-

Gudger, E. W. 1930. Pug-headedness in the striped sea bass, *Roccus lineatus*, and in other related fishes. Bull. Amer. Mus. Nat. Hist., 61(1):1-19.

Guillard, R. R. L. 1975. Culture of phytoplankton for feeding marine invertebrates. *In* Culture of Marine Invertebrate Animals (W. L. Smith and M. H. Chanley, eds.). Plenum Press, New York and London. pp. 29-60.

Hall, R. A., E. G. Zook, and G. M. Meaburn. 1978. National Marine Fisheries Service Survey of Trace Elements in the Fishery Resource. NOAA Tech. Rept. NMFS SSRF-721. 313 pp.

Hallowing Point Field Station. 1976. Potomac Estuary fisheries study - ichthyoplankton and juvenile investigations. Final Report. CEES Rept. No. 76-12 CBL, Maryland Power Plant Siting Program.

Halver, J. E. (ed.). 1972. Fish Nutrition. Academic Press, New York and London. 713 pp.

Hamer, P. E. 1966. The occurrence of striped cusk-eel *Rissola marginata*, in the body cavity of striped bass, *Roccus saxatilis*. Ches. Sci., 7(4):214-215.

Hamer, P. E. 1971. Migratory patterns of some New Jersey striped bass, *Morone saxatilis*. N.J. Dept. of Environmental Protection. Miscellaneous Rept. No. 6M:1-23.

Hansen, L. G., W. B. Wiekhorst, and J. Simon. 1976. Effects of dietary Aroclor 1242 on channel catfish (*Ictalurus punctatus*) and the selective accumulation of PCB components. J. Fish. Res. Board Canada, 33:1343-1352.

Harper, J. L., and R. Jarman. 1972. Investigation of striped bass, *Morone saxatilis* (Walbaum), culture in Oklahoma. Proc. of 25th Ann. Conf. Southeastern Assoc. of Game & Fish Comm.:501-512.

Harper, J. L., R. Jarman, and J. T. Yacovino. 1968. Food habits of young striped bass, *Roccus saxatilis* (Walbaum), in culture ponds. Proc. 22nd Ann. Conf. Southeastern Assoc. Game & Fish Comm.:373-380.

Harrell, R. M., H. A. Loyacono, and J. D. Bayless. 1977. Zooplankton availability and feeding selectivity of fingerling striped bass. Ga. J. Sci., 35:129-135.

Hassler, W. W. 1958. Striped bass in relation to the multiple use of the Roanoke River, N.C. Trans. 23rd N. Amer. Wildl. Conf.:378-391.

Hassler, W. W., and W. T. Hogarth. 1970. The status, abundance and exploitation of striped bass in the Roanoke River and Albemarle Sound, North Carolina, and spawning of striped bass in the Tar River, North Carolina. NOAA-72053115. 346 pp. (COM-72-11379, NTIS)

Hawke, J. P. 1975. Parasite and disease investigations on striped bass and pompano cultured in earthen ponds in south Alabama. Auburn Univ. Dept. Fish. and Allied Aquaculture, SE Coop. Fish Disease Project Newsletter No. 11:3.

Hawke, J. P. 1976. A survey of the diseases of striped bass *Morone saxatilis*, and pompano, *Trachinotus carolinus*, cultured in earthen ponds. Proc. 7th Ann. World Mariculture Society: 495-510.

Hazel, C. R., W. Thomsen, and S. J. Meith. 1971. Sensitivity of striped bass and stickleback to ammonia in relation to temperature and salinity. Calif. Fish & Game, 57(3):138-153.

Heinle, D. R., D. A. Flemer, and J. F. Ustach. 1976. Contribution of tidal marshlands to mid-Atlantic estuarine food chains. *In* Estuarine Processes, Vol. II, Circulation, Sediments, and Transfer of Material in the Estuary (M. Wiley, ed.). Academic Press, New York. p. 309-320.

Helfrich, P., et al. 1973. The feasibility of brine shrimp production on Christmas Island. Sea Grant Technical Rept., UNIHI-SEA-GRANT-TR-73-02. 173 pp.

Heit, M. 1979. Variability of the concentrations of seventeen trace elements in the muscle and liver of a single striped bass, *Morone saxatilis*. Bull. Environm. Contam. Toxicol., 23:1-5.

Helmboldt, C. F., and D. S. Wyand. 1971. Nephroblastoma in a striped bass. J. Wildl. Dis., 7(3):162-165.

Heubach, W., R. J. Toth, and A. M. McCready. 1963. Food of young of the year striped bass (*Roccus saxatilis*) in the Sacramento-San Joaquin River system. Calif. Fish & Game, 49(4):224-239.

Hickey, C. R., Jr. 1976. Fish hematology, its uses and significance. New York Fish & Game J., 23:170-175.

Hiltron, J. W. 1974. Serum transferrin phenotypes in striped bass, *Morone saxatilis*, from the Hudson River. Ches. Sci., 15(4):246-247.

Hoar, W. S., and D. J. Randall (eds.). 1969-1979. Fish Physiology. Vols. 1-8. Academic Press, New York, San Francisco and London.

Hogan, T. M., and B. S. Williams. 1976. Occurrence of the gill parasite, Ergasilus labracis, on striped bass, white perch, and tomcod in the Hudson River. N.Y. Fish & Game J., 23(1):97.

Holland, B. F., and G. F. Yelverton. 1973. Distribution and biological studies of anadromous fishes offshore North Carolina. Div. of Comm. and Sports Fish., N.C. Dept. of Nat. & Econ. Res., Spec. Sci. Rept. No. 24. 132 pp.

Hollis, E. H. 1952. Variations in the feeding habits of the striped bass, Roccus saxatilis (Walbaum), in Chesapeake Bay. Bing. Oceanog. Coll. Bull., 14(1):111-131.

Hollis, E. H. 1967. Investigation of striped bass in Maryland. Md. Dept. od Game & Inland Fish., Md. F-003-R-12. Final Rept. 96 pp.

Holton, M. G. 1874. Letters of Spencer F. Baird. In The progress of fish-culture in the United States by James W. Milner. Rept. U.S. Fish Comm. for 1872-1873, pt. 2, Appendix D: 553-554.

Houde, E. D., and A. J. Ramsay. 1971. A culture system for marine fish larvae. Progr. Fish-Cult., 33(3):156-157.

Hughes, J. S. 1968. Toxicity of pollutants to striped bass. La. Wildlife and Fisheries Commission. Progress Rept., F-015-R-01. 2 pp.

Hughes, J. S. 1969. Toxicity of pollutants to striped bass. La. Wildlife and Fisheries Commission. Progress Rept., F-015-R-02. 3 pp.

Hughes, J. S. 1971. Tolerance of striped bass, Morone saxatilis (Walbaum), larvae and fingerlings to nine chemicals used in pond culture. Proc. of the 24th Ann. Conf. Southeastern Assoc. of Game & Fish Comm.:431-438.

Hughes, J. S. 1973. Acute toxicity of thirty chemicals to striped bass (Morone saxatilis). Presented at the Western Assoc. of State Game & Fish Comm. in Salt Lake City, Utah, July 1973. 15 pp.

Hughes, J. S. 1975. Striped bass, Morone saxatilis (Walbaum), culture investigations in Louisiana with notes on sensitivity of fry and fingerlings to various chemicals. Louisiana Wildlife and Fisheries Comm. Fish. Bull. No. 13. 46 pp.

Humphries, E. T. 1966. Spawning grounds of the striped bass, Roccus saxatilis (Walbaum), in the Tar River, North Carolina. M.S. Thesis, East Carolina College, Greenville, N.C. 50 pp.

Humphries, E. T. 1971. Culture of striped bass (Morone saxatilis, Walbaum) fingerlings in Virginia. Ph.D. Thesis, Virginia Polytechnic Inst. and State University. 77 pp.

Humphries, E. T., and K. B. Cumming. 1973. An evaluation of striped bass fingerling culture. Trans. Amer. Fish. Soc., 102(1):13-20.

Hunn, J. B., and P. F. Robinson. 1966. Some blood chemistry values for five Chesapeake Bay area fishes. Ches. Sci., 7:173-175.

Institute of Environmental Medicine, New York University Medical Center. 1976a. The effects of changes in hydrostatic pressure on some Hudson River biota. Progress Report for 1975. Prepared for Consolidated Edison of New York, Inc. 109 pp.

Institute of Environmental Medicine, New York University Medical Center. 1976b. The effects of temperature and chlorine on Hudson River organisms. Progress Report for 1975. Prepared for Consolidated Edison Co. of New York, Inc. 120 pp.

Institute of Environmental Medicine, New York University Medical Center. 1977. Hudson River ecosystem studies. Effects of entrainment by the Indian Point Power Plant on biota in the Hudson River estuary. Progress Rept. for 1975 for Consolidated Edison Co. of New York, Inc. 355 pp.

Jackson, H. W., and R. E. Tiller. 1952. Preliminary observations on spawning potential in the striped bass (*Roccus saxatilis*, Walbaum). Md. Dept. Res. & Ed., Ches. Biol. Lab. Publ. No. 93. 16 pp.

Janssen, W. A., and C. D. Meyers. 1967. Antibody against an antigen in beef heart muscle found in striped bass *Roccus saxatilis* serum and absent in white perch *Roccus americanus*. Ches. Sci., 8(1):66.

Jessop, B. M., and W. G. Doubleday. 1976. Creel survey and biological study of the striped bass fishery of the Annapolis River, 1975. Dept. of the Environment, Halifax, N.S., Tech. Rept. Ser. No. MAR/T-76-3. 47 pp.

Jessop, B. M., and C. Vithayasai. 1979. Creel surveys and biological studies of the striped bass fisheries of the Shubenacadie, Gaspereau, and Annapolis Rivers, 1976. Dept. of Fish. and Oceans, Halifax, N.S. Fish. and Mar. Serv. Manuscript Rept. No. 1532. 32 pp.

Johns Hopkins University. 1974. Power Plant Site Evaluation. Interim Report. Douglas Point Site. Prepared for Power Plant Siting Program, JHU, PPSE 4-1, vol. 1 & 2.

Johns Hopkins University, Applied Physics Laboratory. 1975. Testimony on striped bass entrainment by Summit Power Station. In the matter of: Delmarva Power and Light Co., Philadelphia Electric Co., Summit Power Stations Units 1 and 2 Dockets Nos. 5-450 and 50-451. 14 March. 98 pp. and appendices.

Johns Hopkins University. 1976. Power Plant Site Evaluation. Final Report - Douglas Point. Prepared for Md. Power Plant Siting Program JHU, PPSE 4-2, Vol. 1, part 1.

Johnson, C. A., and R. Harkema. 1970. A preliminary report on the life history of Pomphorhynchus rocci, an Acanthocephalan parasite of the striped bass. J. Elisha Mitchell Sci. Soc., 86(4):184-185.

Johnson, W. C., and A. J. Calhoun. 1952. Food habits of California striped bass. Calif. Fish & Game, 38(4):531-534.

Jones, K. 1971. Pesticide monitoring. Quarterly report for the period July-September, 1971. Calif. FW-001-R-09. 7 pp.

Jordan, D. S., and C. H. Eigenman. 1890. A review of the genera and species of Serranidae found in the waters of America and Europe. Bull. U.S. Fish Comm. for 1888, 8:329-433.

Kelley, J. R., Jr. 1969. Investigations on the propagation of the striped bass, Morone saxatilis (Walbaum). Ph.D. Thesis, Auburn University, Auburn, Mass. 102 pp. (University Microfilms, Ann Arbor, Michigan, Order No. 71-4010.)

Kelly, R., and H. K. Chadwick. 1971. Some observations on striped bass temperature tolerances. Calif. Dept. Fish and Game, Anad. Fish. Br., Admin. Rept. 71-9. 11 pp.

Kerby, J. H. 1972. Feasibility of artificial propagation and introduction of hybrids of the Morone complex into estuarine environments, with a meristic and morphometric description of the hybrids. Ph.D. Thesis, University of Virginia, Charlottesville. 172 pp. (University Microfilms, Ann Arbor, Michigan, Order No. 72-33, 243.)

Kernehan, R. J. 1974. Ichthyoplankton. In Ecological Studies in the Vicinity of the Proposed Summit Power Station. April 1973 through December 1973. Vol. II (revised). 303 pp.

Kerr, J. E. 1953. Studies on fish preservation at the Contra Costa Steam Plant of the Pacific Gas and Electric Company. Calif. Fish & Game, Fish. Bull. No. 92:1-66.

Kingsford, E. 1975. Treatment of Exotic Marine Fish Diseases. Pet Reference Series No. 1. The Palmetto Publishing Co., St. Petersburg, Florida. 92 pp.

Klontz, G. W. 1973. Syllabus of Fish Health Management. I. Fish Culture Methods. II. Fish Disease Diagnosis. Texas A&M University, Sea Grant #74-401. 165 pp.

Klyshtorin, L. B., and A. A. Yarzhombek. 1975. Some aspects of the physiology of the striped bass, Morone saxatilis. J. of Ichthyol., 15(6): 985-989.

Kohlenstein, L. C. 1981. On the proportion of Chesapeake Bay stock of striped bass that migrates into the coastal fishery. Trans. Amer. Fish. Soc., 110(1):168-179.

Kohlhorst, D. W. 1973. An analysis of the annual striped bass die-off in the Sacramento-San Joaquin Estuary, 1971-1972. Calif. Dept. of Fish & Game, Anad. Fish. Br., Admin. Rept. No. 73-7. 20 pp.

Koo, T. S. Y. 1970. The striped bass fishery in the Atlantic states. Ches. Sci., 11(2):73-93.

Koo, T. S. Y., and M. L. Johnston. 1978. Larva deformity in striped bass, *Morone saxatilis* (Walbaum), and blueback herring, *Alosa aestivalis* (Mitchill), due to heat shock treatment of developing eggs. Environ. Pollut., 16:137-149.

Koo, T. S. Y., and J. S. Wilson. 1972. Sonic tracking striped bass in the Chesapeake and Delaware Canal. Trans. Amer. Fish. Soc., 101(3):453-462.

Korn, S., and R. Earnest. 1974. Acute toxicity of twenty insecticides to striped bass, *Morone saxatilis*. Calif. Fish & Game, 60(3):128-131.

Korn, S., N. Hirsch, and J. W. Struhsaker. 1976. Uptake, distribution, and depuration of 14C-benzene in northern anchovy, *Engraulus mordax*, and striped bass, *Morone saxatilis*. U.S. Fish. Bull, 74(3):545-551.

Korn, S., and D. Macedo. 1973. Determination of fat content in fish with a nontoxic noninflammable solvent. J. Fish. Res. Bd. Canada, 30(12): 1880-1881.

Korn, S., J. W. Struhsaker, and P. Benville, Jr. 1976. Effects of benzene on growth, fat content, and caloric content of striped bass, *Morone saxatilis*. U.S. Fish Bull., 74(3):694-698.

Kornegay, J. W., and E. T. Humphries. 1976. Spawning of the striped bass in the Tar River, North Carolina. Proc. Ann. Conf. Southeastern Assoc. Game Fish Comm., 29:317-325.

Krantz, G. E. 1970. Lymphocystis in striped bass, *Roccus saxatilis*, in Chesapeake Bay. Ches. Sci., 11(2):137-139.

Krouse, J. S. 1968. Effects of dissolved oxygen, temperature, and salinity on survival of young striped bass, *Roccus saxatilis* (Walbaum). M.S. Thesis, University of Maine, Orono. 61 pp.

Kruger, R. L., and R. W. Brocksen. 1978. Respiratory metabolism of striped bass, *Morone*)*saxatilis* (Walbaum), in relation to temperature. J. exp. mar. Biol. Ecol., 31:55-66.

Kumpf, H. E. 1977. Economic impact of the effects of pollution on the coastal fisheries of the Atlantic and Gulf of Mexico regions of the United States of America. FAO Fisheries Technical Paper No. 172. 79 pp.

Lal, K., R. Lasker, and A. Kuljis. 1977. Acclimation and rearing of striped bass larvae in sea water. Calif. Fish & Game, 63(4):210-218.

Lasker, R. 1974. Induced maturation and spawning of marine fish at the Southwest Fisheries Center, LaJolla, California. Proceedings World Mariculture Society, 5:313-318.

Laurence, G. C. 1977. Caloric values of some North Atlantic calanoid copepods. U.S. Fish. Bull., 75:218-220.

Lawler, J. P. 1972. The effect of entrainment at Indian Point on the population of the Hudson River striped bass. Testimony before the United States Atomic Energy Commission in the Matter of Consolidated Edison Company of New York, Inc. (Indian Point Station, Unit No. 2). Docket No. 50-247, April 5, 1972.

Lawler, J. P., R. A. Norris, G. Goldwyn, K. A. Abood, and T. L. Englert. 1974. Hudson River striped bass life cycle model. In Entrainment and Intake Screening. Proc. 2nd Workshop on Entrainment and Intake Screening (L. D. Jensen, ed.). The Johns Hopkins University, Baltimore, MD. pp. 83-94.

Lawler, Matusky and Skelly Engineers. 1974. 1973 Hudson River Aquatic Ecology Studies - Bowling Point and Lovell Generating Station. Report to Orange and Rockland Utilities, Inc., December. 5 vols.

Lawler, Matusky and Skelly Engineers. 1975. Report on development of a real-time, two dimensional model of the Hudson River striped bass population. Consolidated Edison Company of New York, Inc. LMS Project No. 115-49. 69 pp. + tables.

Leidy, J. 1888. Parasites of the striped bass. Proc. Acad. Nat. Sci. Philad., 2:125-128.

Leim, A. H., and W. B. Scott. 1968. Fishes of the Atlantic Coast of Canada. Bull. Fish. Res. Bd. Canada No. 155. 485 pp.

Lewis, R. M. 1957. Comparative study of populations of the striped bass. U.S. Fish & Wildl. Serv., Spec. Sci. Rept. Fish No. 204. 54 pp.

Lewis, R. M. 1961. Comparison of three tags on striped bass in the Chesapeake Bay area. Ches. Sci., 2(1&2):3-8.

Lewis, R. M. 1962. Sexual maturity as determined from ovum diameters in striped bass from North Carolina. Trans. Amer. Fish. Soc., 91:279-282.

Lewis, R. M., and R. R. Bonner, Jr. 1966. Fecundity of striped bass (Roccus saxatilis Walbaum). Trans. Amer. Fish. Soc., 95(3):328-331.

Lewis, W. M., R. C. Heidinger, and B. L. Tetzlaff. 1977. Striped bass rearing experiments, 1976. Prepared for Consolidated Edison Co. of New York, Inc. University of Southern Illinois, Carbondale. May. 197 pp.

Lindsay, J. A., and R. L. Moran. 1976. Relationships of parasitic isopods *Lironeca ovalis* and *Olencira praegustator* to marine fish biota in Delaware River. Trans. Amer. Fish. Soc., 105(2):327-332.

Linton, E. 1898. Notes on trematode parasites of fishes. Proc. U.S. Nat. Mus., 20:507-548.

Linton, E. 1901. Parasites of fishes of the Woods Hole region. Bull. U.S. Fish Comm., 19:405-492.

Linton, E. 1924. Notes on cestode parasites of sharks and skates. Proc. U.S. Nat. Mus., 64(art. 21):1-114.

Loeber, T. S. 1951. A report of an investigation of the temperature and salinity relationships of striped bass and salmon in connection with the Reber plan. Rept. Bur. of Fish Conserv., Calif. Dept. Fish & Game, Inland Fish. Br., No. 51-27. 40 pp.

Lovett, R. J., W. H. Gutenmann, I. S. Pakkala, W. D. Youngs, D. J. Lisk, G. E. Burdick, and E. J. Harris. 1972. A survey of the total cadmium content of 406 fish from 49 New York fresh waters. J. Fish. Res. Bd. Canada, 29(9):1283-1290.

Luhning, C. W. 1973. Residues of MS-222, benzocaine, and their metabolites in striped bass following anesthesia. U.S. Dept. of Interior, Bur. Sport Fish. & Wildl. Investigations in Fish Control No. 52. 11 pp.

Lund, W. A., Jr. 1957. Morphometric study of the striped bass, *Roccus saxatilis*. U.S. Fish & Wildl. Serv., Spec. Sci. Rept.-Fish. No. 216: 1-24.

Lyman, H. 1961. A sixteen pound pugheaded striped bass from Massachusetts. Ches. Sci., 2:101-102.

Mahoney, J. B., F. H. Midlige, and D. G. Deuel. 1973. A fin rot disease of marine and euryhaline fishes in the New York Bight. Trans. Amer. Fish. Soc., 102(3):596-605.

Manooch, C. S. III. 1973. Food habits of yearling and adult striped bass, *Morone saxatilis* (Walbaum), from Albemarle Sound. North Carolina. Ches. Sci., 14(2):73-86.

Mansueti, R. 1958. Eggs, larvae and young of the striped bass, *Roccus saxatilis*. Ches. Biol. Lab. Contr. No. 112:1-35.

Mansueti, R. 1960. An unusually large pugheaded striped bass, *Roccus saxatilis* from Chesapeake Bay, Maryland. Ches. Sci., 1(2):111-113.

Mansueti, R. 1961. Age, growth and movements of the striped bass, *Roccus saxatilis*, taken in size selective fishing gear in Maryland. Ches. Sci., 2(1&2):9-36.

Mansueti, R. 1962. Effects of civilization on striped bass and other estuarine biota in Chesapeake Bay and tributaries. Proc. 14th Ann. Gulf & Caribbean Inst. (Univ. of Miami): 110-136.

Mansueti, R., and G. Murphy. 1961. Further returns of striped bass, Roccus saxatilis, tagged from deep water during winter in Chesapeake Bay, Maryland. Ches. Sci. 2(3-4):209-212.

Marcy, B. C., Jr., and R. C. Galvin. 1973. Winter-spring sport fishery in the heated discharge of a nuclear power plant. J. Fish. Biol., 5: 541-547.

Markert, C. L. and I. Faulhaber. 1965. Lactate dehydrogenase isoenzyme patterns of fish. J. Exp. Zool., 159:319-332.

Markle, D. F. 1976. The seasonality of availability and movements of fishes in the channel of the York River, Virginia. Ches. Sci., 17(1):50-55.

Markle, D. G., and G. C. Grant. 1970. The summer food habits of young-of-the-year striped bass in three Virginia rivers. Ches. Sci., 11(1): 50-54.

Mason, H. W. 1882. Report of operations in the Navesink River, New Jersey, in 1879 , in collecting living striped bass for transportation to California. Rept. U.S. Fish Comm. for 1879, 7:663-666.

Massmann, W. H., and A. L. Pacheco. 1961. Movements of striped bass in Virginia waters of Chesapeake Bay. Ches. Sci., 2(1-2):37-44.

McCoy, E. G. 1959. Quantitative sampling of striped bass, Roccus saxatilis (Walbaum), eggs in the Roanoke River, North Carolina. M.S. Thesis, North Carolina State University, Raleigh. 136 pp.

McFadden, J. T. (ed.). 1977a. Influence of Indian Point Unit 2 and other steam electric generating plants on the Hudson River Estuary, with emphasis on striped bass and other fish populations. Submitted to Consolidated Edison Co. of New York, Inc., January.

McFadden, J. T. 1977b. An argument supporting the reality of compensation in fish populations and a plea to let them exercise it. In Proceedings of the Conference on Assessing the Effects of Power-Plant-Induced Mortality on Fish Populations (W. Van Winkle, ed.). Pergamon Press, N.Y. pp. 153-183.

McFadden, J. T., and J. P. Lawler (eds.) 1977. Supplement I to Influence on Indian Point Unit 2 and other steam electric generating plants on the Hudson River Estuary, with emphasis on striped bass and other fish populations. Submitted to Consolidated Edison Co. of New York, Inc., July.

McFarland, W. N., and G. W. Klontz. 1969. Anesthesia in fishes. Chapter 26 of Anesthesia in Laboratory Animals. Federation Proceedings, Vol. 28(4):1535-1540.

McHugh, J. J., and R. C. Heidinger. 1977. Effects of light on feeding and egestion time of striped bass fry. Prog. Fish-Cult., 39:33-34.

McHugh, J. J., and R. C. Heidinger. 1978. Effects of light shock on striped bass fry. Prog. Fish-Cult., 40:82.

McHugh, J. L. 1972. Marine fisheries of New York State. U.S. Fish. Bull., 70(3):585-610.

McIlwain, T. D. 1974. Experimental stocking of striped bass. Completion Report, 1 September 1970-31 August 1973, AFCS-4. 64 pp.

McIlwain, T. D. 1975. Striped bass rearing and stocking program - Mississippi. Annual Progress Report, 1 September 1973 to 31 August 1974, AFCS-5-1. 48 pp.

McKechnie, R. J., and L. W. Miller. 1971. The striped bass party boat fishery: 1960-1968. Calif. Fish & Game, 57(1):4-16.

Mehrle, P. M., F. L. Mayer, and W. W. Johnson. 1977. Diet quality in fish toxicology: Effects on acute and chronic toxicity. In Aquatic Toxicology and Hazard Evaluation (F.L. Mayer and J.L. Hamelink, eds.). American Society for Testing and Materials, Pahiladelphia, PA. Special Technical Publication 634:269-280.

Meldrim, J. W., and J. T. Gift. 1971. Temperature preference, avoidance and shock experiments with estuaring fishes. Ichthyological Associates, Inc. Bull. 7. 75 pp.

Meldrim, J. W., J. J. Gift, and B. R. Petrosky. 1974. The effect of temperature and chemical pollutants on the behavior of several estuarine organisms. Ichthyological Associates, Inc. Bull. 11. 129 pp.

Merriman, D. 1937. Notes on the life history of the striped bass, Roccus lineatus. Copeia, 1:15-36.

Merriman, D. 1940. The osteology of the striped bass (Roccus saxatilis) Annals and Mag. Nat. Hist., Series 11(5):55-64.

Merriman, D. 1941. Studies on the striped bass (Roccus saxatilis) of the Atlantic Coast. U.S. Fish & Wildl. Serv., Fish. Bull., 50(35):1-77.

Merriner, J. V., and W. J. Hogman. 1973. Feasibility of increasing striped bass populations by stocking under-utilized nursery grounds. Completion Report AFS - 6-3, July 1, 1970-September 30, 1973. 196 pp.

Meshaw, J. C., Jr. 1969. A study of feeding selectivity of striped bass fry and fingerlings in relation to zooplankton availability. M.S. Thesis, North Carolina State University, Raleigh. 58 pp.

Meyer, F. P., and J. A. Robinson. 1973. Branchiomycosis: A new fungal disease of North American fishes. Progr. Fish-Cult., 35(2):74-77.

Meyerhoff, R. D. 1975. Acute toxicity of benezene, a component of crude oil, to juvenile striped bass (Morone saxatilis). J. Fish. Res. Bd. Canada, 32(10):1864-1866.

Middaugh, D. P., J. A. Couch, and A. M. Crane. 1977. Responses of early life history stages of the striped bass, Morone saxatilis to chlorination. Ches. Sci., 18(1):141-153.

Miller, L. W. 1974. Mortality rates for California striped bass (Morone saxatilis) from 1965-1971. Calif. Fish & Game, 60(4):157-171.

Miller, L. W. 1977. An evaluation of sampling nets used for striped bass and Neomysis in the Sacramento-San Joaquin Estuary. Calif. Fish & Game, Anad. Fish. Br., Admin. Rept. No. 77-3. 29 pp.

Miller, P. E. 1977. Experimental study and modeling of striped bass egg and larval mortality. Ph.D. Thesis, The Johns Hopkins University, Baltimore. 199 pp. (University Microfilms, Ann Arbor, Michigan, Order No. 77-7749.)

Miller, P. E. 1978. Food habit study of striped bass post yolk-sac larvae. Prepared by Chesapeake Bay Institute, The Johns Hopkins University. Special Report 68 (Ref. No. 78-8), November. 49 pp.

Mitchill, S. L. 1814. Report in part of Samual L. Mitchill, M.D., Professor of Natural History, etc., of the fishes of New York. D. Carlisle, New York: 1-30. (From a reprint edited by Theodore N. Gill, Washington, 1898.)

Mitchill, S. L. 1815. The fisheries of New York, described and arranged. Trans. Lit. Philos. Soc. N.Y., 1:355-492.

Moore, C. J., and D. T. Burton. 1974. Movements of striped bass, Morone saxatilis, tagged in Maryland waters of Chesapeake Bay. Trans. Amer. Fish. Soc., 104(4):703-709.

Morgan, A. R., and A. R. Gerlach. 1950. Striped bass studies on Coos Bay, Oregon in 1949 and 1950. Oregon Fish Comm., Contr. No. 14:1-31.

Morgan, R. P. II. 1971. Comparative electrophoretic studies on the striped bass, Morone saxatilis, and white perch, M. americana, and electrophoretic identification of five populations of striped bass in the upper Chesapeake Bay. Ph.D. Thesis, University of Maryland, College Park. 104 pp. (University Mircrofilms, Ann Arbor, Michigan, Order No. 71-25,968.)

Morgan, R. P. II. 1975. Distinguishing larval white perch and striped bass by electrophoresis. Ches. Sci., 16(1):68-70.

Morgan, R. P. II, T. S. Y. Koo, and G. E. Kranz. 1973. Electrophoretic determination of populations of the striped bass, Morone saxatilis, in the upper Chesapeake Bay. Trans. Amer. Fish. Soc., 102(1):21-32.

Morgan, R. P. II, and V. J. Rasin, Jr. 1973. Effects of salinity and temperature on the development of eggs and larvae of striped bass and white perch. Appendix X to Hydrographic and Ecological Effects of Enlargement of the Chesapeake and Delaware Canal. Contract No. DACW-61-71-C-0062, Army Corps of Engineers, Philadelphia District. NRI Ref. No. 73-109. 37 pp.

Morgan, R. P. II, V. J. Rasin, Jr., and L. A. Noe. 1973. Effects of suspended sediments on the development of eggs and larvae of striped bass and white perch. Appendix XI to Hydrographic and Ecological Effects of Enlargement of the Chesapeake and Delaware Canal. Contract No. DACW-61-71-C-0062, Army Corps of Engineers, Philadelphia District. NRI Ref. No. 73-110. 21 pp.

Morgan, R. P. II, R. E. Ulanowicz, V. J. Rasin, Jr., and L. A. Noe. 1976. Effects of shear on striped bass and white perch eggs and larvae. Trans. Amer. Fish. Soc., 105(1):145-154.

Morgan, R. P. II, and R. W. Prince. 1977. Chlorine toxicity to eggs and larvae of five Chesapeake Bay fishes. Trans. Amer. Fish. Soc., 106(4): 380-385.

Moss, F. T. 1974. Successful Striped Bass Fishing. International Marine Publishing Co. 208 pp.

Murawski, W. S. 1958. Comparative study of populations of the striped bass, Roccus saxatilis (Walbaum), based on lateral-line scale counts. M.S. Thesis, Cornell University, Ithaca, N.Y. 80 pp.

Murawski, W. S. 1969. A study of the striped bass, Morone saxatilis, foulhooking problem in New Jersey waters. New Jersey Dept. of Conservation and Economic Development, Div. of Fish and Game, Bureau of Fisheries, Nacote Creek Research Station, Misc. Rept. No. 4M. 40 pp.

Murphy, G. J. 1960. Availability of striped bass during summers of 1958 and 1959 as reflected in commercial haul seine catch. Ches. Sci., 1(1):74-75.

Nash, C. E. 1973. Automated mass-production of Artemia salina nauplii for hatcheries. Aquaculture, 2:289-298.

National Research Council. 1973. Nutrient Requirements of Trout, Salmon, and Catfish. National Academy of Sciences, Washington, D.C. 57 pp.

National Research Council. 1977. Nutrient Requirements of Warmwater Fishes. National Academy of Sciences, Washington, D.C. 78 pp.

Nelson, N. C. 1974. A review of the literature on the use of malachite green in fisheries. U.S. Fish and Wildlife Service, Bureau of Sport Fisheries and Wildlife. PB-235 450. 79 pp.

Nichols, P. R. 1962. Atlantic coast striped bass program. *In* Ann. Rept., Bur. Comm. Fish. Biol. Lab., Beaufort, N.C., for the fiscal year ending June 30, 1962. U.S. Fish & Wildl. Ser., Circular 184:17-18.

Nichols, P. R., and R. V. Miller. 1967. Seasonal movements of striped bass, *Roccus saxatilis* (Walbaum) tagged and released in the Potomac River, Maryland, 1959-61. Ches. Sci., 8(2):102-124.

Norny, E. R. 1882. A proposed pond for rearing striped bass (*Roccus lineatus*) in Delaware Bay. Bull. U.S. Fish Comm. for 1881, 1:260-261.

O'Connell, C. P. 1976. Histological criteria for diagnosing the starving condition in early post yolk sac larvae of the northern anchovy, *Engraulis mordax* Girard. J. exp. mar. Biol. Ecol., 25:285-312.

O'Connor, J. M., and S. A. Schaffer. 1977. The effects of sampling gear on the survival of striped bass ichthyoplankton. Ches. Sci., 18(3):312-315.

O'Mally, M., and J. Boone. 1972. Oxygen vital to normal hatching and survival in striped bass. Maryland Fish and Wildlife News, 3(2):1.

O'Rear, C. W., Jr. 1971. Some environmental influences on the zinc and copper content of striped bass, *Morone saxatilis* (Walbaum). Ph.D. Thesis, Virginia Polytechnic Institute and State University, Blacksburg. 78 pp.

Orsi, J. J. 1970. A comparison of scales, otoliths, and operculae in striped bass aging. The Resources Agency, Dept. of Fish and Game, California, Anad, Fish. Admin. Rept. No. 70:15. 5 pp.

Orsi, J. J. 1971. The 1965-1967 migrations of the Sacramento-San Joaquin estuary striped bass population. Calif. Fish & Game, 57(4):257-267.

Otto, R. S. 1975. Isoenzyme systems of the striped bass (*Morone saxatilis*) and congeneric perichthyid fishes. Ph.D. Thesis, University of Maine, Orono. 71 pp. (University Microfilms, Ann Arbor, Michigan, Order No. 76-7437.)

Otwell, W. S., and J. V. Merriner. 1975. Survival and growth of juvenile striped bass, *Morone saxatilis*, in a factorial experiment with temperature, salinity, and age. Trans. Amer. Fish. Soc., 104(3):560-566.

Overstreet, R. M. 1971. *Neochasmus sogandaresi* n. sp. (Trematoda: Cryptogonimidae) from striped bass in Mississippi. Trans. Amer. Microsc. Soc., 90(1):87-89.

Oviatt, C. A. 1977. Menhaden, sport fish and fishermen. University of Rhode Island Marine Technical Report No. 60. 24 pp.

Paffenhofer, G. A. 1967. Caloric content of larvae of the brine shrimp *Artemia salina*. Helg. Wiss. Meer., 16:130-135.

Pakkala, I. S., M. N. White, D. J. Lisk, G. E. Burdick, and E. J. Harris. 1972. Arsenic content of fish from New York State waters. N.Y. Fish and Game J., 19(1):12-31.

Pandian, T. J. 1970. Intake and conversion of food in the fish *Limanda limanda* exposed to different temperatures. Marine Biol., 5:1-17.

Paperna, I., and D. E. Zwerner. 1974. *Kudoa cerebralis* sp. n. (Myxosporidea, Chloromyxidae) from the striped bass, *Morone saxatilis* (Walbaum). J. Protozool., 21(1):19-25.

Paperna, I., and D. E. Zwerner. 1976a. Parasites and diseases of striped bass, *Morone saxatilis* (Walbaum), from the lower Chesepeake Bay. J. Fish Biol., 9:267-287.

Paperna, I., and D. E. Zwerner. 1976b. Studies on *Ergasilus labracis* Kroyer (Cyclopidea: Ergasilidae) parasite on striped bass, *Morone saxatilis*, from the lower Chesapeake Bay. I. Distribution, life cycle and seasonal abundance. Can. J. Zool., 54(4):449-462.

Pearson, J. C. 1933. Movements of striped bass in Chesapeake Bay. Maryland Fisheries, 22:15-17.

Pearson, J. C. 1938. The life history of the striped bass, or rockfish, *Roccuss saxatilis* (Walbaum). U.S. Bur. of Fish. Bull., 28(49):825-851.

Peddicord, R. K., V. A. McFarland, D. P. Belfiori, and T. E. Byrd. 1975. Effects of suspended solids on San Francisco Bay organisms. *In* Dredge Disposal Study, San Francisco Bay and Estuary, Appendix G, Physical Impact Study. 158 pp. + appendices.

Peddicord, R. K., and V. A. McFarland. 1978. Effects of suspended dredged material on aquatic animals. Prepared for Office, Chief of Engineers, U.S. Army (under DMRP Work Unit No. 1D09). Tech. Rept. D-78-29. 102 pp. + appendices.

Perlmutter, A., E. Leff, E. E. Schmidt, R. Heller, and M. Sicliano. 1967a. Distribution and abundance of fishes along the shores of the lower Hudson during the summer of 1966. *In* Hudson River Ecology (First Symposium), Hudson River Valley Commission, New York: 147-200.

Perlmutter, A., E. E. Schmidt, and E. Leff. 1967. Distribution and abundance of fish along the shores of the lower Hudson River during the summer of 1965. N.Y. Fish and Game Jour., 14(1):47-75.

Persoone, G., and P. Sorgeloos. 1975. Technological improvements for the cultivation of invertebrates as food for fishes and crustaceans. I. Devices and methods. Aquaculture, 6:275-289.

Peterson, E. J., and R. C. Robinson. 1967. A meal-gelatin diet for aquarium fishes. Progr. Fish-Cult., 29:170-171.

Pfunderer, H. A., S. S. Talmadge, B. N. Collier, W. Van Winkle, Jr., and C. P. Goodyear. 1975. Striped bass — a selected, annotated bibliography. Oak Ridge National Laboratory, Environmental Sci. Div. Pub. No. 615. 158 pp.

Phillips, A. M., Jr. 1969. Nutrition, digestion, and energy utilization. In Fish Physiology (W. S. Hoar and D. J. Randell, eds.). Academic Press, New York. Vol. 1, pp. 391-432.

Phillips, A. M., Jr., R. S. Nielsen, and D. R. Brockway. 1954. A comparison of hatchery diets and natural food. Progr. Fish-Cult., 16:153-157.

Pickford, G. E., and J. W. Atz. 1957. The Physiology of the Pituitary Gland of Fishes. New York Zoological Society, New York. 613 pp.

Plosila, D. S. 1961. Lower Susquehanna River sport fishery survey, 1957-1960. In The Susquehanna Fishery Study, 1957-1960 (R. R. Whitman, Proj. Leader). Md. Dept. Res. and Educ., Contrib. 169:55-76.

Polgar, T. T. 1977. Striped bass ichthyoplankton abundance, mortality, and production estimation of the Potomac River populations. In Proceedings of the Conference on Assessing the Effects of Power-Plant-Induced Mortality on Fish Populations (W. Van Winkle, ed.). Pergamon Press, N.Y. pp. 110-126.

Polgar, T. T., R. E. Ulanowicz, and D. A. Pyne. 1975. Preliminary analyses of physical transport and related striped bass ichthyoplankton distribution properties in the Potomac River in 1974. Potomac River Fisheries Program Report Series. Ref. No. PRFP-75-2. February 1975. 51 pp.

Powell, M. R. 1973. Cage and raceway culture of striped bass in brackish water in Alabama. Proc. 26th Ann. Conf. Southeastern Assoc. Game & Fish Comm., pp. 345-356.

Powell, M. R. 1976. Two years of brackish water pond culture of striped bass (Morone saxatilis) in south Alabama. Proceedings World Mariculture Society, 7:93-97.

Radovich, J. 1963. Effect of ocean temperature on the seaward movements of striped bass, Roccus saxatilis, on the Pacific Coast. Calif. Fish & Game, 49(3):191-206.

Radtke, L. D. 1966. Distribution and abundance of adult and subadult striped bass (*Roccus saxatilis*), in the Sacramento-San Joaquin Delta. *In* Ecological studies of the Sacramento-San Joaquin estuary, Part II. Calif. Dept. Fish & Game, Fish Bull. No. 136:15-27.

Radtke, L. D., and J. L. Turner. 1967. High concentrations of total dissolved solids block spawning migration of striped bass, *Roccus saxatilis*, in the San Joaquin River, California. Trans. Amer. Fish. Soc. 96(4):405-407.

Radtke, R. L. 1978. Aberrant sagittae from larval striped bass, *Morone saxatilis*. Copeia, 1978(4):712-713.

Raney, E. C. 1952. The life history of the striped bass (*Roccus saxatilis*, Walbaum). Bull. Bingham Oceanogr. Coll., 14(1):5-97.

Raney, E. C. 1957. Subpopulations of the striped bass *Roccus saxatilis* (Walbaum) in tributaries of Chesapeake Bay. *In* Contributions to the Study of Subpopulations of Fishes. U.S. Fish and Wildl. Serv., Spec. Sci. Rept.-Fish. No. 208:85-107.

Raney, E. C., and D. P. DeSylva. 1953. Racial investigations of the striped bass, *Roccus saxatilis* (Walbaum). Jour. Wildl. Mgt., 17:495-509.

Raney, E. C., and E. C. Weller. 1972. Two decades of study of the striped bass, *Morone saxatilis* (Walbaum). Proc. 15th Ann. International Game Fish Res. Conf., December 1972: 28-70.

Raney, E. C., and W. S. Woolcott. 1955. Races of the striped bass, *Roccus saxatilis* (Walbaum), in southeastern United States. Jour. Wildl. Mgt., 19:444-450.

Raney, E. C., W. S. Woolcott, and A. G. Mehring. 1954. Migratory pattern and racial structure of Atlantic coast striped bass. Trans. 19th N. Amer. Wildl. Conf., pp. 376-396.

Rathjen, W. F., and L. C. Miller. 1957. Aspects of the early life history of the striped bass (*Roccus saxatilis*) in the Hudson River. N.Y. Fish & Game Jour. 4(1):43-60.

Ray, R. H., and L. J. Wirtanen. 1970. Striped bass *Morone saxatilis* (Walbaum), 1969 report on the development of essential requirements for production. U.S. Fish and Wildl. Serv., Div. of Hatcheries Publ. 46 pp.

Raymont, J. E. G., J. Austin, and E. Linford. 1963. Biochemical studies on marine zooplankton. I. The biochemical composition of *Neomysis integer*. J. Cons. Inter. Explor. Mer., 28(1):354-363.

Redpath, G. S. 1972. Temperature and its effect on the growth and production of juvenile striped bass (Morone saxatilis). M.S. Thesis, University of California, Davis. 37 pp.

Rees, R. A. 1974. Propagation of striped bass. Final Report. Ga. Game & Fish Div., Statewide Fish. Investig. F-31-5, Study XIV: 14-62.

Reeves, W. C. 1972a. The effects of increased water hardness, source of fry and age of stocking on the survival of striped bass, Morone saxatilis (Walbaum), fry and effects of two feeding regimes and sources of fingerlings on survival and production of advanced fingerling striped bass in ponds. M.S. Thesis, Auburn University, Auburn, Alabama. 58 pp.

Reeves, W. C. 1972b. Striped bass disease problems. Auburn University Dept. Fish. and Allied Aquaculture. S.E. Cooperative Disease Project Newsletter No. 9: 3.

Reeves, W. C., and J. F. Germann. 1972. Effects of increased water hardness, source of fry and age at stocking on survival of striped bass fry in earthen ponds. Proc. 25th Ann. Conf. Southeastern Assoc. of Game and Fish Comm., pp. 542-548.

Regan, D. M., T. L. Wellborn, Jr., and R. G. Bowker. 1968. Striped bass Roccus saxatilis (Walbaum) development of essential requirements for production. U.S. Fish and Wildl. Serv., Bur. Sport Fish. and Wildl. Div. of Fish Hatcheries, Atlanta, Georgia. 133 pp.

Rehwoldt, R., G. Bida, and B. Nerrie. 1971. Acute toxicity of copper, nickel and zinc ions to some Hudson River fish species. Bull. Environ. Contam. and Toxicol., 6(5):445-448.

Rehwoldt, R. E., E. Kelley, and M. Mahoney. 1977. Investigations into the acute toxicity and some chronic effects of selected herbicides and pesticides on several fresh water fish species. Bull. Environ. Contam. and Toxicol., 18(3):361-365.

Rehwoldt, R. E., W. Mastrianni, E. Kelley, and J. Stall. 1978. Historical and current heavy metal residues in Hudson River fish. Bull. Environ. Contamin. and Toxicol., 15:335-339.

Resources Management Associates. 1979. A review and analysis of the Lawler, Matusky and Skelly real time life cycle model of the Hudson River striped bass population. Prepared for the Federal Energy Regulatory Commission, Washington, D.C. 146 pp.

Rhodes, W., and J. V. Merriner. 1973. A preliminary report on closed system rearing of striped bass sac fry to fingerling size. Progr. Fish-Cult., 35(4):199-201.

Ribelin, W. E., and G. Migaki (eds.). 1975. The Pathology of Fishes. The University of Wisconsin Press, Madison, Wisconsin. 1004 pp.

Richards, C. E. 1962. A survey of salt-water sport fishing in Virginia, 1955-1960. Ches. Sci., 3(4):223-235.

Ricker, W. E. (ed.). 1971. Methods of Assessment of Fish Production in Fresh Waters. IBP Handbook No. 3. Blackwell Scientific Publications, Oxford. 384 pp.

Rinaldo, R. G. 1971. *Morone saxatilis* and *Morone americanus* spawning and nursery area in the York-Pamunkey River. M.S. Thesis, Virginia Institute of Marine Science, Gloucester Point, Virginia. 64 pp.

Ritchie, D. E., Jr. 1965. Sex determination of live striped bass *Roccus saxatilis* (Walbaum) by biopsy technique. Ches. Sci., 6(3):141-145.

Ritchie, D. E., Jr. 1970. Evaluation of gonadal biopsy technique in striped bass based on tagged fish returns. Ches. Sci., 11(4):210-215.

Robinson, J. B. 1960. The age and growth of striped bass (*Roccus saxatilis*) in California. Calif. Fish & Game, 46(3):279-290.

Rogers, B. A. 1978. Temperature and the rate of early development of striped bass, *Morone saxatilis* (Walbaum). Ph.D. Thesis, University of Rhode Island, Kingston. 193 pp. (University Microfilms, Ann Arbor, Michigan, Order No. 78-13271.)

Rogers, B. A., and D. T. Westin. 1979. The combined effects of temperature and delayed feeding on the survival and growth of larval striped bass, *Morone saxatilis* (Walbaum). *In* Advances of Marine Environmental Research, Proceedings of a Symposium (F. S. Jacoff, ed.), June 15-16, 1977. Narragansett, R.I. EPA 600/9-79-035:234-250.

Rogers, B. A., and D. T. Westin. 1981. Laboratory studies on the effects of temperature and delayed initial feeding on the development of striped bass larvae. Trans. Amer. Fish. Soc., 110(1):100-110.

Rogers, B. A., D. T. Westin, and S. B. Saila. 1977. Life stage duration in Hudson River striped bass. University of Rhode Island Marine Technical Report 31. 111 pp.

Rogers, R. D., and D. E. Stevens. 1971. Distribution of young striped bass (*Morone saxatilis*) in the Sacramento-San Joaquin Delta at Collinsville and Pottsburg. Calif. Dept. Fish & Game, Anad. Fish. Br. Admin. Rept. 71-12. 14 pp.

Rosko, M. 1966. Secrets of Striped Bass Fishing. Macmillan Co., N.Y. 238 pp.

Ryder, J. A. 1887. *Roccus lineatus* (Bloch) Gill: (The striped bass, or rockfish.) Hybridization of the striped bass with other fishes. *In* On the development of osseous fishes, including marine and fresh-water forms. Rept. U.S. Fish Comm. for 1885, 13:502-505.

Saila, S. B., and E. Lorda. 1977. Sensitivity analysis applied to a matrix model of the Hudson River striped bass population. In Proceedings of the Conference on Assessing the Effects of Power-Plant-Induced Mortality on Fish Populations (W. Van Winkle, ed.). Pergamon Press, New York. pp. 311-332.

Sandoz, N. O., and K. H. Johnston. 1966. Culture of striped bass *Roccus saxatilis* (Walbaum). Proc. 19th Ann. Conf. Southeastern Assoc. Game and Fish Comm., pp. 390-394.

Saski, S. 1966a. Distribution of young striped bass (*Roccus saxatilis*), in the Sacramento-San Joaquin Delta. In Ecological studies of the Sacramento-San Joaquin estuary. Part II. Calif. Dept. Fish & Game, Fish Bull. No. 136:44-58.

Sasaki, S. 1966b. Distribution of juvenile striped bass, *Roccus saxatilis*, in the Sacramento-San Joaquin Delta. Calif. Dept. Fish & Game, Fish. Bull. No. 136:59-67.

Sazaki, M. W. Heubach, and J. E. Skinner. 1972. Some preliminary results on the swimming ability and impingement tolerance of young-of-the-year steelhead trout, king salmon and striped bass. Final Report, Anadromous Fisheries Act Project, Calif. AFS-13. 34 pp.

Schaefer, R. H. 1967. Species composition, size and seasonal abundance of fish in the surf waters of Long Island. N.Y. Fish & Game Jour., 14(1): 1-46.

Schaefer, R. H. 1968a. Size, age composition and migration of striped bass from the surf waters of Long Island. N.Y. Fish & Game Jour., 15(1): 1-51.

Schaefer, R. H. 1968b. Sex composition of striped bass from the Long Island surf. N.Y. Fish & Game Jour., 15(1):117-118.

Schaefer, R. H. 1970. Feeding habits of striped bass from the surf waters of Long Island. N.Y. Fish & Game Jour., 17(1):1-17.

Schaefer, R. H. 1972. A short-range forecast function for predicting the relative abundance of striped bass in Long Island waters. N.Y. Fish & Game Jour., 19(2):178-181.

Schnick, R. A. 1973. Formalin as a therapeutant in fish culture. U.S. Fish & Wildlf. Ser., Bur. of Sport Fisheries and Wildl. PB-237 198. 131 pp.

Schubel, J. R. 1974. Effects of exposure to time-excess temperature histories typically experienced at power plants on the hatching success of fish eggs. Special Report 32, PPRP-4 of Chesapeake Bay Institute of The Johns Hopkins University, Baltimore, Md. 37 pp.

Schultz, L. P. 1931. Hermaphroditism in the striped bass. Copeia, 2:64.

Scofield, E. C. 1931. The striped bass of California (Roccus lineatus). Calif. Div. Fish & Game, Fish Bull. No. 29:1-82.

Scofield, N. B. 1910. Notes on the striped bass in California. 21st Biennial Rept. Bd. of Fish & Game Comm. of Calif. for 1909-1910:104-109.

Scofield, N. B., and G. A. Coleman. 1910. Notes on spawning and hatching of striped bass eggs at Bouldin Island hatchery. 21st Biennial Rept. Calif. Bd. of Fish & Game Comm. for 1909-1910:109-117.

Scruggs, G. D., Jr. 1957. Reproduction of resident striped bass in Santee-Cooper reservoir, South Carolina. Trans. Amer. Fish. Soc., 85:144-159.

Scruggs, G. D., Jr., and J. C. Fuller, Jr. 1955. Indications of a freshwater population of striped bass, Roccus saxatilis (Walbaum), in Santee-Cooper reservoir. Proc. of the Southeastern Assoc. of Game & Fish Comm., pp. 64-69.

Shannon, E. H. 1970. Effect of temperature changes upon developing striped bass eggs and fry. Proc. of the 23rd Ann. Conf. Southeastern Assoc. Game & Fish Comm., pp. 365-274.

Shannon, E. H., and W. B. Smith. 1967. Preliminary observations of the effect of temperature on striped bass eggs and sac fry. Proc. 21st Ann. Conf. Southeastern Assoc. Game & Fish Comm., pp. 257-260.

Shapovalov, L. 1936. Food of the striped bass. Calif. Fish & Game, 22(4): 261-270.

Shearer, L. W., D. E. Ritchie, Jr., and C. M. Frisbie. 1962. Sport fishing survey in 1960 of the lower Patuxent estuary and the 1958 year class of striped bass. Ches. Sci., 3(1):1-17.

Shebley, W. H. 1927. History of fish planting in California. Calif. Fish & Game, 13:163-174.

Shuster, C. N. 1959. A biological evaluation of the Delaware River Estuary. University of Delaware Marine Lab., Information Series, Publ. No. 3: 1-77.

Sidell, B. D., R. G. Otto, and D. A. Powers. 1978. A biochemical method for distinction of striped bass and white perch larvae. Copeia, 1978(2):340-343.

Sidell, B. D., R. G. Otto, D. A. Powers, M. Karweit, and J. Smith. 1980. Apparent genetic hemogeneity of spawning striped bass in the upper Chesapeake Bay. Trans. Amer. Fish. Soc., 109(1):99-107.

Sidwell, V. D., P. R. Foncannon, N. S. Moore, and J. C. Bonnet. 1974. Composition of the edible portion of raw (fresh or frozen) crustaceans, finfish, and mollusks. I. Protein, fat, moisture, ash, carbohydrate, energy value, and cholesterol. Marine Fisheries Review, 36(3):21-35.

Slobodkin, L. B., and S. Richman. 1961. Calories/gm in species of animals. Nature (Lond.), 191:299.

Smith, H. M. 1896. A review of the history and results of the attempts to acclimatize fish and other water animals in the Pacific states. Bull. U.S. Fish Comm. for 1895, 15:379-472.

Smith, L. D. 1970. Life history studies of striped bass. Final Report, Anadromous Fish Project, Georgia, AFS-2. 134 pp.

Smith, T. I. J., J. S. Jopkins, and P. A. Sandifer. 1978. Development of a large-scale *Artemia* hatching system utilizing recirculated water. Proceedings World Mariculture Society, 9:701-714.

Smith, W. B., W. B. Bonner, and B. L. Tatum. 1967. Premature egg procurement from striped bass *Roccus saxatilis*. Proc. 20th Ann. Conf. Southeastern Assoc. Game & Fish Comm., pp. 324-330.

Snieszko, S. F., G. L. Bullock, E. Hollis, and J. G. Boone. 1964. *Pasteurella* sp. from an epizootic of white perch (*Roccus americanus*) in Chesapeake Bay tidewater areas. J. Bacteriology, 88(6):1813-1814.

Snyder, J. P. 1913. Notes on striped bass. Trans. Amer. Fish. Soc., 43:93-96.

Snyder, J. P. 1915. Efforts to ripen striped bass, 1915. Trans. Amer. Fish. Soc., 45(1):40-45.

Solorzano, L. 1969. Determination of ammonia in natural waters by the phenolhypochlorite method. Limnol. Oceanog., 14:799-801.

Sommani, P. 1972. A study on the population dynamics of striped bass (*Morone saxatilis*, Walbaum) in the San Francisco Bay estuary. Ph.D. Thesis, University of Washington, Seattle. 145 pp. (University Microfilms, Ann Arbor, Michigan, Order No. 72-20, 893.)

Sorgeloos, P., and G. Persoone. 1975. Technological improvements for the cultivation of invertebrates as food for fishes and crustaceans. II. Hatching and culturing of the brine shrimp, *Artemia salina* L. Aquaculture, 6:303-317.

Sorgeloos, P., G. Persoone, M. Baeza-Mesa, E. Bossuyt, and E. Bruggeman. 1978. The use of *Artemia* cysts in aquaculture: The concept of "hatching efficiency" and description of a new method for cyst processing. Proceedings World Mariculture Society, 9:715-721.

Spagnoli, J. J., and L. C. Skinner. 1977. PCB's in fish from selected waters of New York State. Pesticide Monitoring Journ., 11(2):69-87.

Spotte, S. 1979. Fish and Invertebrate Culture. Water Management in Closed Systems. 2nd Edition. Wiley-Interscience Publication, John Wiley and Sons, New York. 179 pp.

Starks, E. C. 1901. Synonymy of the fish skeleton. Proc. Acad. Sci. Wash., 3:507-539, 3 pls.

Stevens, D. E. 1966. Food habits of striped bass, Roccus saxatilis, in the Sacramento-San Joaquin Delta. Part II. Calif. Dept. Fish & Game, Fish Bull., 136:68-96.

Stevens, D. E. 1977a. Striped bass (Morone saxatilis) monitoring techniques in the Sacramento-San Joaquin Estuary. In Proceedings of the Conference on Assessing the Effects of Power-Plant-Induced Mortality on Fish Populations (W. Van Winkle, ed.). Pergamon Press, N.Y. pp. 91-109.

Stevens, D. E. 1977b. Striped bass (Morone saxatilis) year class strength in relation to river flow in the Sacramento-San Joaquin Estuary, California. Trans. Am. Fish. Soc., 106(1):34-42.

Stevens, R. E. 1958. The striped bass of the Santee-Cooper reservoir. Proc. 11th Ann. Conf. Southeastern Assn. of Game & Fish Comm., pp. 253-264.

Stevens, R. E. 1965. A final report on the use of hormones to ovulate striped bass, Roccus saxatilis (Walbaum). 18th Ann. Conf. Southeastern Assoc. Game & Fish Comm. 21 pp.

Stevens, R. E. 1966. Hormone-induced spawning of striped bass for reservoir stocking. Progr. Fish-Cult., 28(1):19-28.

Stevens, R. E. 1967. Striped bass rearing. Cooperative Fisheries Unit, N.C. State University, Raleigh. 14 pp.

Stolte, L. W. 1974. Tag returns for striped bass tagged in New Hampshire. Underwater Natur., 8(1):26-31.

Struhsaker, J. S., D. Y. Hashimoto, S. M. Girard, F. T. Prior, and T. D. Cooney. 1973. Effect of antibiotics on survival of carangid fish larvae (Caranx mate), reared in the laboratory. Aquaculture, 2:53-88.

Surber, E. W. 1958. Results of striped bass (Roccus saxatilis) introduction into freshwater impoundments. Proc. 11th Ann. Conf. Southeastern Assoc. Game & Fish Comm., pp. 273-276.

Swartzman, G., R. Deriso, and C. Cowan. 1977. Comparison of simulation models used in assessing the effects of power-plant-induced mortality on fish populations. In Proceedings of the Conference on Assessing the Effects of Power-Plant-Induced Mortality on Fish Populations (W. Van Winkle, ed.). Pergamon Press, N.Y., pp. 333-361.

Tagatz, M. E. 1961. Tolerance of striped bass and American shad to changes of temperature and salinity. U.S. Fish & Wildl. Serv., Spec. Sci. Rept.-Fish. No. 338. 8 pp.

Talbot, G. B. 1967. Teratological notes on striped bass Roccus saxatilis of San Francisco Bay. Copeia, 1967(2):459-461.

Tatum, B. L., J. Bayless, E. G. McCoy, and W. B. Smith. 1965. Preliminary experiments in artificial propagation of striped bass. 19th Ann. Conf. Southeastern Assoc. Game & Fish Comm., pp. 374-389.

Texas Instruments Inc. 1973. Hudson River ecology study in the area of Indian Point. First Annual Report to Consolidated Edison Co. of N.Y., Inc. 348 pp. + 3 appendices.

Texas Instruments Inc. 1974a. Hudson River ecological study in the area of Indian Point. 1973 Annual Report. Prepared for Consolidated Edison Co. of N.Y., Inc. July 1974.

Texas Instruments Inc. 1974b. Acute and chronic effects of evaporative cooling tower blowdown and power plant chemical discharges on white perch (Morone americana) and striped bass (M. saxatilis). Prepared for Consolidated Edison Co. of N.Y., Inc. November 1974. 39 pp.

Texas Instruments Inc. 1975a. First Annual Report for the Multiplant Impact Study of the Hudson River Estuary. Prepared under contract to Consolidated Edison Co. of N.Y., Inc. Jointly financed by Consolidated Edison Co. of N.Y., Inc., Orange and Rockland Utilities, Inc., and Central Hudson Gas and Electric Corp., July 1975, Vol. I & II.

Texas Instruments Inc. 1975b. Final report of the synopic subpopulation analysis, Phase I: Report on the feasibility of using innate tags to identify striped bass (Morone saxatilis) from various spawning rivers. Prepared under contract with Consolidated Edison Co. of N.Y., Inc., September, 1975.

Texas Instruments Inc. 1975c. Hudson River ecological study in the area of Indian Point. 1974 Annual Report. Prepared for Consolidated Edison Co. of N.Y., Inc.

Texas Instruments Inc. 1976a. Fisheries Survey of the Hudson River. March-December 1973, Vol. IV. Prepared for Consolidated Edison Co. of N.Y., Inc. June 1976.

Texas Instruments Inc. 1976b. Hudson River ecological study in the area of Indian Point. Thermal effects report preapred for Consolidated Edison Company of N.Y., Inc. September, 1976.

Texas Instruments Inc. 1976c. Hudson River ecological study in the area of Indian Point. 1975 Annual Report. Prepared for Consolidated Edison Company of New York, Inc. December.

Texas Instruments Inc. 1977a. Feasibility of culturing and stocking Hudson River striped bass. 1975 Annual Report. Prepared for Consolidated Edison Co. of N.Y., Inc. April.

Texas Instruments Inc. 1977b. 1974 year-class report for the Multiplant Impact Study of the Hudson River Estuary. Prepared under contract with Consolidated Edison Co. of N.Y., Inc., Orange and Rockland Utilities, Inc., and Central Hudson Gas and Electric Corp. May. 3 Vols.

Texas Instruments Inc. 1977c. Feasibility of culturing and stocking Hudson River striped bass. An overview, 1973-1975. Prepared for Consolidated Edison Co. of N.Y., Inc. June.

Theilacker, G. H. 1978. Effect of starvation on the histological and morphological characteristics of jack mackerel, *Trachurus symmetricus*, larvae. U.S. Fish. Bull., 76(2):403-414.

Thomas, J. L. 1967. The diet of juvenile and adult striped bass, *Roccus saxatilis*, in the Sacramento-San Joaquin River system. Calif. Fish & Game, 53(1):49-62.

Tiller, R. E. 1942. Indications of compensatory growth in the striped bass, *Roccus saxatilis* Walbaum, as revealed by a study of the scales. M.S. Thesis, University of Maryland, College Park. 36 pp. (Published with same title in Ches. Biol. Lab. Publ. No. 57:1-16.)

Tiller, R. E. 1950. A five-year study of the striped bass fishery of Maryland, based on analyses of the scales. Chesapeake Biol. Lab. Publ. No. 85. 30 pp.

Tong, S. C., W. H. Gutenmann, D. J. Lisk, G. E. Burdick, and E. J. Harris. 1972. Trace metals in New York State fish. N.Y. Fish & Game Jour., 19(2):123-131.

Townes, H. K., Jr. 1937. Studies on the food organisms of fish. *In* A biological survey of the lower Hudson watershed. Suppl. 26th Rept. N.Y. St. Conserv. Comm., 1936:225-226.

Trent, W. L. 1962. Growth and abundance of young-of-year striped bass, *Roccus saxatilis* (Walbaum) in Albemarle Sound, North Carolina. M.S. Thesis, North Carolina State University, Raleigh. 66 pp.

Trent, W. L., and W. W. Hassler. 1968. Gill net selection, migration, size and age composition, sex ratio, harvest efficiency, and management of striped bass in the Roanoke River, North Carolina. Ches. Sci., 9(4): 217-232.

Tresselt, E. F. 1952. Spawning grounds of the striped bass, *Roccus saxatilis* (Walbaum) in Virginia. Bull. Bingham Oceanog. Coll., 14(1): 98-110.

Turner, J. L. 1976. Striped bass spawning in the Sacramento and San Joaquin Rivers in central California from 1963 to 1972. Calif. Fish & Game, 62(2):106-118.

Turner, J. L., and H. K. Chadwick. 1972. Distribution and abundance of young-of-the-year striped bass, Morone saxatilis, in relation to river flow in the Sacramento-San Joaquin estuary. Trans. Amer. Fish. Soc., 101(3):442-452.

Turner, J. L., and T. C. Farley. 1971. Effects of temperature, salinity, and dissolved oxygen on the survival of striped bass eggs and larvae. Calif. Fish & Game, 57(4):268-273.

Turner, J. L., and D. W. Kelly (compilers). 1966. Ecological studies of the Sacramento-San Joaquin Delta. Part II - Fishes of the Delta. Calif. Fish & Game, Fish. Bull. No. 136:1-168.

U. S. Fish and Wildlife Service. 1904/05-. Propagation and distribution of food fishes. Report year irregular. 1904/05-1929/30 issued as Bureau of Fisheries Document; 1930/31-1939/40 as U.S. Bureau of Fisheries Administration Report; 1940/41-1961/62 as U.S. Fish and Wildlife Service Statistical Digest; and 1967- as Fish Distribution Report of Division of Hatcheries. 1912/13-1939-40 issues as Appendix to U.S. Bureau of Fisheries Report of the Commissioner of Fisheries.

U. S. Fish and Wildlife Service. 1941-. Fishery Statistics of the United States. Issue complied for each year as Fish and Wildl. Serv. Statistical Digest; and yearly by areas as Current Fishery Statistics (CFS) series.

U. S. Senate. 1979. Hearings before the subcommittee on resource protection of the Committee on Environment and Public Works. United States Senate 96th Congress on S. 838. Serial No. 96-H7. 168 pp.

United States Nuclear Regulatory Commission. 1975. Final Environmental Statement related to operation of Indian Point Nuclear Generating Plant Unit No. 3. Consolidated Edison Company of New York, Inc. Docket No. 50-296. February 1975. Vols. I & II.

Valenti, R. J., J. Aldred, and J. Liebell. 1976. Experimental marine cage culture of striped bass in northern waters. Proceedings World Mariculture Society, 7:99-108.

Van Winkle, W., B. W. Rust, C. P. Goodyear, S. R. Blum, and P. Thall. 1974. A striped-bass population model and computer programs. Oak Ridge National Laboratory, Contract No. W7405-eng-26. Environmental Sciences Division, Publ. No. 643. (ORNL-TM-4578) 200 pp.

Van Winkle, W., S. W. Christensen, and G. Kauffman. 1976. Critique and sensitivity analysis of the compensation function used in the LMS Hudson River striped bass models. Environ. Sci. Div., Publ. No. 944, Oak Ridge National Laboratory. (ORNL/TM-5437) 100 pp.

Van Winkle, W., S. W. Christensen, and J. S. Suffern. 1979a. Incorporation of sublethal effects and indirect mortality in modeling population-level impacts of a stress, with an example involving power-plant entrainment and striped bass. Oak Ridge National Laboratory, Environ. Sci. Div. Publ. No. 1295. (NUREG/CR-0638; ORNL/NUREG/TM-288) 24 pp.

Van Winkle, W. B., L. Kirk, and B. W. Rust. 1979b. Periodicities in Atlantic coast striped bass (*Morone saxatilis*) commercial fisheries data. J. Fish. Res. Bd. Canada, 36:54-62.

Vincent, W. S., H. O. Halvorson, H. R. Chen, and D. Shin. 1969. A comparison of ribosomal gene amplification in uni- and multi-nucleolate oocytes. Exp. Cell Res., 57(2&3):240-250.

Vladykov, V. D. 1957. Fish tags and tagging in Quebec waters. Trans. Amer. Fish. Soc., 86:345-349.

Vladykov, V. D., and D. H. Wallace. 1938. Is the striped bass (*Roccus lineatus*) of Chesapeake Bay a migratory fish? Trans. Amer. Fish. Soc., 67:67-86.

Vladykov, V. D., and D. H. Wallace. 1952. Studies of striped bass, *Roccus saxatilis* (Walbaum), with special reference to the Chesapeake Bay region during 1936-1938. Bull. Bingham Oceanog. Coll., 14(1):132-177.

Wales, J. H. 1946. Fungus in air bladder of striped bass. Calif. Fish & Game, 32:31.

Wallace, D. N. 1975. A critical comparison of the biological assumptions of Hudson River striped bass models and field survey data. Trans. Amer. Fish. Soc., 104(4):710-717.

Wallace, D. N. 1978. Two anomalies of fish larval transport and their importance in environmental assessment. N.Y. Fish & Game Journ., 25(1):59-71.

Ware, F. J. 1971. Some early life history of Florida's inland striped bass, *Morone saxatilis*. Proc. 24th Ann. Conf. Southeastern Assoc. Game & Fish Comm., pp. 439-447.

Warsh, K. L. 1975. Hydrological-biological models of the impact of entrainment of spawn of the striped bass (*Morone saxatilis*) in proposed power plants at two areas in the upper Chesapeake Bay. The Johns Hopkins University, Applied Physics Laboratory. PPSE-T-1: 1-94 + appendices.

Wawronowicz, L. J., and W. M. Lewis. 1979. Evaluation of the striped bass as a pond-reared food fish. Progr. Fish-Cult., 41(3):138-140.

Wedemeyer, G. A., and W. T. Yasutake. 1977. Clinical methods for the assessment of the effects of environmental stress on fish health. U.S. Dept. of Interior, Fish and Wildlife Service, Technical Papers, 89:1-18.

Wellborn, T. L., Jr. 1969. The toxicity of nine therapeutic and herbicidal compounds to striped bass. Prog. Fish-Cult., 31(1):27-32.

Wellborn, T. L., Jr. 1971. Toxicity of some compounds to striped bass fingerlings. Prog. Fish-Cult., 33(1):32-36.

Westin, D. T. 1978. Serum and blood from adult striped bass, Morone saxatilis. Estuaries, 1(2): 123-125.

Whitehead, P. J. P., and A. C. Wheeler. 1966. The generic names used for sea basses of Europe and North America (Pices: Serranidae). Ann. Mus. Civ. Stor. Nat. Genoa, 76:23-41.

Wigfall, M., and J. M. Barkuloo. 1976. A preliminary report on the abundance and biology of stocked striped bass in the Choctawhatchee River System, Florida. Proc. Ann. Conf. Southeastern Assoc. Game Fish. Comm., 29:152-161.

Williams, H. M. 1972. Preliminary fecundity studies of the hybrid (striped bass X white bass) in two South Carolina reservoirs. Proc. 25th Ann. Conf. Southeastern Assoc. Game & Fish Comm., pp. 536-542.

Williamson, F. A. 1974. Population studies of striped bass (Morone saxatilis) in the Saint John and Annapolis Rivers. M.S. Thesis, Acadia University, Wolfville, N.S., Canada. 60 pp.

Wilson, J. S., R. P. Morgan II, P. W. Jones, H. R. Lunsford, Jr., J. Lawson, and J. Murphy. 1976. Potomac River fishery study - striped bass spawning stock assessment. Final Rept., 1975. Ches. Biol. Lab UMCEES Ref. No. 76-14. 61 pp + tables.

Windom, H., R. Stickney, R. Smith, D. White, and F. Taylor. 1973. Arsenic, cadmium, mercury, and zinc in some species of North Atlantic finfish. J. Fish. Res. Bd. Canada, 30(2):275-279.

Wirtanen, L. J., and R. H. Ray. 1971. Striped bass, Morone saxatilis (Walbaum). 1970 Report on the development of essential requirements for production. U.S. Fish and Wildl. Serv., Div. Hatcheries Publ. 37 pp.

Wolke, R. L. 1975. A survey of the diseases and parasites of fishes of the Thames River Watershed, Connecticut. In An Evaluation of the Fishery Resources of the Thames River Watershed, Connecticut (R. L. Hames, ed.). Storrs Agric. Expt. Sta., The University of Connecticut, Storrs, Bull., 435:74-100.

Wolke, R. E., D. S. Wyand, and W. H. Khairallah. 1970. A light and electron microscopic study of epitheliocystis disease in the gills of Connecticut striped bass (Morone saxatilis) and white perch (Morone americanus). J Comp. Pathol., 80(4):559-563.

Wood, G., and L. Hintz. 1971. Lipid changes associated with the degradation of fish tissue. J. Official Assoc. of Anal. Chem., 54(5):1019-1023.

Preceding page blank

Woolcott, W. S. 1957. Comparative osteology of serranid fishes of the genus Roccus (Mitchill). Copeia, 1957(1):1-10.

Worth, S. G. 1882. The artificial propagation of the striped bass (Roccus lineatus) on Albemarle Sound. Bull. U.S. Fish Comm. for 1881, 1:174-177.

Worth, S. G. 1904. The recent hatching of striped bass, and possibilities with other commercial species. Trans. Amer. Fish. Soc., 33:223-230.

Worth, S. G. 1910. Progress of hatching striped bass. Trans. Amer. Fish. Soc., 39:155-159.

Worth, S. G. 1912. Fresh-water and angling grounds for the striped bass. Trans. Amer. Fish. Soc., 41:115-126.

Yarzhombek, A. A., and L. B. Klyashiorin. 1974. The relationship between standard metabolism and resting metabolism in fishes. J. Ichthyl., 14(3):439-443.

Zawacki, C. S., and P. T. Briggs. 1976. Fish investigations in Long Island Sound at a nuclear power station site at Shoreham, N.Y. Fish & Game J., 23(1):34-50.

*

Finlayson, B. J., and D. E. Stevens. 1977. Mortality-temperature relationships for young striped bass (Morone saxatilis) entrained at two power plants in the Sacramento-San Joaquin Delta, California. Calif. Fish & Game, Anad. Fish. Br. Admin. Rept. No. 77-6:22 pp.

Sills, J. B., and P. D. Harman. 1971. Efficacy and residues of quinaldine sulfate, an anaesthetic, for striped bass (Roccus saxatilis). Proc. 24th Ann. Conf. Southeastern Assoc. of Game & Fish Comm.:546-549.

Sindermann, C. J. 1979. Pollution-associated diseases and abnormalities of fish and shellfish: A review. U. S. Fish. Bull. 76(4):717-749.